

**A LATE HOLOCENE VEGETATIONAL SEQUENCE
FROM THE SOUTHEAST MISSOURI OZARKS**

A Thesis

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by

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ABSTRACT

Palynological investigations of a peat deposit from a small sinkhole bog, Buttonbush Bog, and two archaeological sites, Round Spring Shelter, Round Spring Site 23SH19, and Gooseneck Site 23CT54 located in Shannon and Carter counties Missouri, provide a vegetational record for the last 3100 years in the southeast Ozarks. The Buttonbush Bog core has a basal radiocarbon date of 3130 ± 100 years B.P. Pollen spectra from the basal zone indicate a mixed oak forest with minor components of pine and hickory. Shortly thereafter, pine becomes more abundant, suggesting the presence of a pine-oak forest that has undergone very little change to the present. A small *Ambrosia* rise indicates land clearance and pioneer settlement about 165 years ago.

The pollen sequence from Round Spring Shelter also suggests the presence of a pine-oak forest in the vicinity of Round Spring. However, the pollen spectra from Gooseneck Site 23CT54 indicate a mixed oak-hickory forest in the locality during the time of Indian occupation. Preliminary analysis of the fossil phytoliths from Gooseneck indicates that four grass subfamilies may have grown at the site.

TABLE OF CONTENTS

	Page
ABSTRACT	i
TABLE OF CONTENTS	ii
LIST OF TABLES	vii
LIST OF FIGURES	viii
LIST OF PLATES	x
ACKNOWLEDGMENTS	xi
INTRODUCTION	1
Previous Investigations	3
Pollen Studies	3
Phytolith Studies	8
The Study Locality: Its Setting	11
Bedrock Geology	15
Precambrian	15
Cambrian	15
Ordovician	17
Mississippian	21
Soils	25
Climate	26
Flora	33
ARCHAEOLOGICAL OVERVIEW OF THE SOUTHEAST MISSOURI OZARKS	38
Unspecialized Hunter-Gatherer Tradition	42
Early Man Period: 12,000 B.C. (?-13,950 B.P.)	42

	Page
Early Hunter Tradition	42
Paleoindian Period: 12,000-8000 B.C. (13,950-9950 B.P.)	42
Hunter-Forager Tradition	45
Dalton Period: 8000-7000 B.C. (9950-8950 B.P.)	45
Forager Tradition	45
Early Archaic Period: 7000-5000 B.C. (8950-6950 B.P.)	45
Middle Archaic Period: 5000-3000 B.C. (6950-4950 B.P.)	46
Late Archaic Period: 3000-1000 B.C. (4950-2950 B.P.)	46
Prairie-Forest Potter Tradition	47
Early Woodland Period: 1000-500 B.C. (2950-2450 B.P.)	47
Middle Woodland Period: 500 B.C.-A.D. 400 (2450-1550 B.P.)	48
Late Woodland Period: A.D. 400-900 (1550-1050 B.P.)	48
Village Farmer Tradition	49
Early Mississippi Period: A.D. 900-1200 (1050-750 B.P.)	49
Middle Mississippi Period: A.D. 1200-1450 (750-500 B.P.)	50
Late Mississippi Period: A.D. 1450-1700 (500-250 B.P.)	51
Historical Period: A.D. 1700-Present (250 B.P.-Present)	51
SPECIFIC SITE DESCRIPTIONS	54
Buttonbush Bog	54
Round Spring Shelter, Round Spring Site 23SH19	54
Gooseneck Site 23CT54	59
Ozark Sink Pond	59
Incidental Sites	61

	Page
METHODS	62
Coring Methods and Collection of Archaeological Sediments	62
Buttonbush Bog	62
Round Spring Shelter, Round Spring Site 23SH19	62
Gooseneck Site 23CT54	62
Ozark Sink Pond	63
Collection of Modern Pollen Rain Samples	63
Pollen Analytical Methods	64
Buttonbush Bog	64
Round Spring Shelter, Round Spring Site 23SH19	64
Gooseneck Site 23CT54	65
Modern Pollen Rain Samples	65
Palynomorph Identification and Counting Techniques	66
Statistical Analysis and Data Processing	67
Diagram Construction	69
Loss-On-Ignition of Organic and Carbonate Carbon	69
Clastic Sediment Analysis	70
Collection of Contemporary Vegetation	72
Plant Identification	72
Contemporary Vegetation	73
Phytolith Identification and Counting Techniques	74
Phytolith Analytical Methods	74
Buttonbush Bog	74
Round Spring Shelter, Round Spring Site 23SH19	75
Gooseneck Site 23CT54	75
Phytolith Key	75

	Page
MODERN POLLEN RAIN	81
SEDIMENT STRATIGRAPHY	83
Buttonbush Bog	83
Round Spring Shelter, Round Spring Site	83
Gooseneck Site 23CT54	89
RADIOCARBON DATES AND SEDIMENTATION RATES	94
Buttonbush Bog	94
POLLEN CONCENTRATION AND INFLUX RATES	97
Buttonbush Bog	97
Round Spring Shelter, Round Spring Site 23SH19	99
Gooseneck Site 23CT54	102
POLLEN DIAGRAMS	105
Buttonbush Bog	105
Zone 1: <i>Quercus</i> Pollen-Assemblage Zone (292-302 cm)	105
Zone 2: <i>Quercus-Carya-Pinus</i> Pollen-Assemblage Zone (17-292 cm)	107
Zone 3: <i>Ambrosia</i> Pollen-Assemblage Zone (0-17 cm)	109
Round Spring Shelter, Round Spring Site 23SH19	110
Gooseneck Site 23CT54	113
22N/4-5E (Samples 1 and 5)	114
22-23N/22-23E (Samples 2 and 6)	115
22-23N/24-25E (Samples 3 and 7)	115
31-32N/12-13E (Sample 4)	116
32-33N/12-13E (Sample 8)	116

	Page
PHYTOLITH ASSEMBLAGES	122
Contemporary Vegetation Reference Samples	122
Buttonbush Bog	122
Round Spring Shelter, Round Spring Site 23SH19	122
Gooseneck Site 23CT54	123
SUMMARY AND CONCLUSIONS	125
REFERENCES CITED	130
APPENDIX I: PLANT NAMES	140
APPENDIX II: CONTEMPORARY VEGETATION	148
A. Contemporary Vegetation Collected at Buttonbush Bog	148
B. Contemporary Vegetation Collected at Ozark Sink Pond	150
C. Contemporary Vegetation Collected at Round Spring Site 23SH19	152
D. Contemporary Vegetation Collected at Gooseneck Site 23CT54	154
E. Contemporary Vegetation Collected at Incidental Sites	157
APPENDIX III: LABORATORY PROCEDURES	158
A. Pollen Preparation Procedure for Small Quaternary Sediment Samples	158
B. Pollen Preparations from Large-Volume Sediment Samples	160
C. Pollen Preparation from Moss Polsters for Modern Pollen Rain Studies	163
D. Laboratory Technique for Determination of Organic Carbon and Carbonate by Loss-On-Ignition	166
E. Microstratigraphic Technique of Grain Size Analysis: A Method to Obtain Percentages of Sand/Silt/Clay	168
F. Extraction of Opal Phytoliths from Peat Samples I	172
G. Extraction of Opal Phytoliths from Sediment Samples V	173
H. Extraction of Opal Phytoliths from Plant Samples VIII	175
APPENDIX IV: PLANTS FROM THE STUDY AREA THAT HAVE BEEN ANALYZED FOR PHYTOLITHS	177
APPENDIX V: POTENTIAL NATIVE FOOD AND BEVERAGE PLANTS OF THE SOUTHEAST MISSOURI OZARKS	193

LIST OF TABLES

	Page
TABLE 1. Summary of the Inferred Dominant Vegetation and Faunal Assemblages from Spring Deposits in the Western Missouri Ozarks	5
TABLE 2. Contemporary Climatic Data for the Study Area	30
TABLE 3. Climatic Episodes and Proposed Chronological Periods Used to Discuss Missouri Archaeology	43
TABLE 4. Cultural Traditions in Relation to Archaeological Periods	44
TABLE 5. Chronological Sequence of Cultural Developments in the Study Area	53
TABLE 6. POLPERC Pollen Program Divisions for Percentages	67
TABLE 7. Grass Classification Scheme— General Categories	77
TABLE 8. Sediment Lithology at Buttonbush Bog	84
TABLE 9. Munsell Color Descriptions for Round Spring Shelter	88
TABLE 10. Sample Locations and Munsell Colors for Gooseneck Site 23CT54	91
TABLE 11. Comparison of Plant Taxa Found at Gooseneck Site 23CT54	118
TABLE 12. Comparison of Ethnobotanical Material and Fossil Pollen from the Same Provenience at Gooseneck Site 23CT54	121
TABLE 13. Distribution Pattern of Grass Silica Phytoliths	123

LIST OF FIGURES

	Page
Figure 1. A map of Missouri showing sites of previous pollen studies and the sites investigated in this study	2
Figure 2. Subprovinces and escarpments of Ozark Plateaus province	12
Figure 3. Geographical provinces of the Ozark Highland of Missouri	14
Figure 4. Precambrian and Cambrian stratigraphy of Shannon and Carter counties	16
Figure 5. Ordovician stratigraphy of Shannon and Carter counties	18
Figure 6. Mississippian stratigraphy of Shannon and Carter counties	23
Figure 7. Distribution of the major soil areas of Missouri	27
Figure 8. Border of Borchert's Climatic Regions 2 and 4	31
Figure 9. Climatic regions of eastern America	32
Figure 10. Principal plant regions of Missouri	34
Figure 11. Map of vegetational associations in Missouri	35
Figure 12. Archaeological-physiographic regions of Missouri	39
Figure 13. Stream drainage localities of Missouri	40
Figure 14. Photograph of Buttonbush Bog	55
Figure 15. Map of Round Spring Shelter, Round Spring Site 23SH19	56
Figure 16. Photograph of Round Spring	57
Figure 17. Map of Round Spring Site 23SH19	58
Figure 18. Map of Gooseneck Site 23CT54	60
Figure 19. Dumbbell	78
Figure 20. Cross	78
Figure 21. Saddle	79
Figure 22. Long trapezoid	79
Figure 23. Short trapezoid	80

	Page
Figure 24. Sediment stratigraphy at Buttonbush Bog	85
Figure 25. Sediment stratigraphy at Round Spring Shelter, Round Spring Site 23SH19	86
Figure 26. Photograph of the probable hearth stone from the Round Spring Shelter core	87
Figure 27. Typical soil profile of the terrace edge, Gooseneck Site 23CT54	90
Figure 28. Gooseneck Site 23CT54 test unit soil profiles for samples collected	92
Figure 29. Sediment stratigraphy at Gooseneck Site 23CT54	93
Figure 30. Radiocarbon dates from the Buttonbush Bog core plotted against depth	95
Figure 31. Pollen sum and total fossil palynomorph concentration (grains/cm ³) plotted against depth for the Buttonbush Bog core	98
Figure 32. Pollen sum and total fossil palynomorph influx (grains/cm ²) plotted against depth for the Buttonbush Bog core	100
Figure 33. Pollen sum and total fossil palynomorph concentration (grains/cm ³) plotted against depth for the Round Spring Shelter core	101
Figure 34. Pollen sum and total fossil palynomorph concentration (grains/cm ³) plotted against level provenience for the Gooseneck Site 23CT54 samples	103
Figure 35. Photograph of the channel cut through the gravel path located on top of the natural bridge at Round Spring during the 1982 flood	112
Figure 36. Summary of the environmental and cultural records found at Buttonbush Bog, Round Spring Shelter, Round Spring Site 23SH19, and Gooseneck Site 23CT54	129

LIST OF PLATES

Plate 1. OZARK MODERN POLLEN RAIN, Percentage Diagram

Plate 2. OZARK MODERN POLLEN RAIN, Percentage Diagram (95% Confidence Intervals)

Plate 3. BUTTONBUSH BOG, Shannon County Missouri, Pollen Percentage Diagram

Plate 4. BUTTONBUSH BOG, Shannon County Missouri, Pollen Concentration Diagram

Plate 5. BUTTONBUSH BOG, Shannon County Missouri, Pollen Influx Diagram

Plate 6. BUTTONBUSH BOG, Shannon County Missouri, Pollen Percentage Diagram
(95% Confidence Intervals)

Plate 7. ROUND SPRING SHELTER, Shannon County Missouri,
Pollen Percentage Diagram

Plate 8. ROUND SPRING SHELTER, Shannon County Missouri,
Pollen Percentage Diagram

Plate 9. ROUND SPRING SHELTER, Shannon County Missouri,
Pollen Percentage Diagram (95% Confidence Intervals)

Plate 10. GOOSENECK SITE 23CT54, Carter County Missouri,
Pollen Percentage Diagram

Plate 11. GOOSENECK SITE 23CT54, Carter County Missouri,
Pollen Concentration Diagram

Plate 12. GOOSENECK SITE 23CT54, Carter County Missouri,
Pollen Percentage Diagram (95% Confidence Intervals)

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INTRODUCTION

In the past, paleobotanical investigations have relied heavily on pollen as the primary indicator of vegetational change. Plant macrofossils have also been used in conjunction with pollen studies (Watts and Winter, 1966; Watts and Bright, 1968; Van Zant, 1976, 1979). As early as 1908, H.C. Schellenberg recognized the importance of phytoliths in paleobotanical investigations in archaeological contexts. However, prior to 1970 very few paleobotanical studies in archaeological context included phytolith analysis (Rovner, 1983). In the 1950s and 1960s soil scientists used phytoliths in a broad paleoecological context to distinguish soil types developed under different vegetational conditions (Baker, 1959; Witty and Knox, 1964; Verma and Rust, 1969). In at least four instances, pollen and phytolith analyses have been undertaken in the same study (Moody, 1972; Bartoli and Guillet, 1977; Schreve-Brinkman, 1978; Kurman, 1981).

The principal objective of this study was the interpretation of the late Holocene vegetational history of the southeast Missouri Ozarks based on palynological data from Buttonbush Bog; Round Spring Shelter, Round Spring Site 23SH19; and Gooseneck Site 23CT54 (Fig. 1). Correlative objectives were to undertake preliminary comparisons between modern plant phytolith assemblages and fossil phytolith assemblages and to compare the pollen and phytolith data from the same sediment samples as well as provide a general overview of the paleoecology and the potential natural resources available in the study locality.

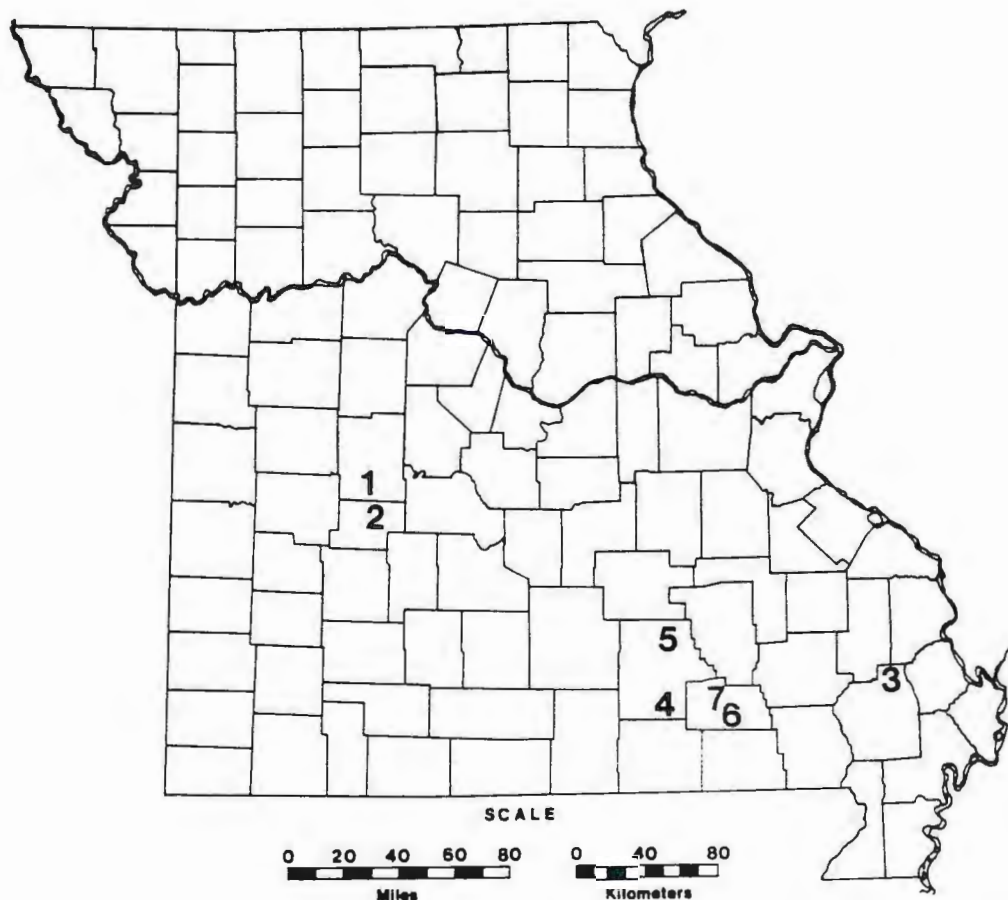


Figure 1. A map of Missouri showing sites of previous pollen studies and sites investigated in this study.

Explanation

1. Boney and Phillips springs
2. Kirby, Jones, Trolinger, and Koch springs
3. Old Field Swamp
4. Buttonbush Bog
5. Round Spring Shelter, Round Spring Site 23SH19
6. Gooseneck Site 23CT54
7. Ozark Sink Pond

PREVIOUS INVESTIGATIONS

Pollen Studies

The first palynological study in Missouri was of Boney Spring (Fig. 1), Benton County (Mehring, Schweger, Wood, and McMillan, 1968). Mehring and others (1968) found a spruce-dominated pollen spectrum and associated *Picea* and *Larix* macrofossils (common names are listed in Appendix I). They interpreted the pollen spectrum as evidence for late Pleistocene boreal forest elements in the Ozark Highlands.

Mehring, King, and Lindsay (1970) examined Boney Spring in more detail and examined Trolinger Spring, Hickory County (Fig. 1). They recognized three pollen-assemblage zones from the combined data of the two sites. The three zones are generally characterized as a NAP-pine zone (NAP = nonarboreal pollen), a spruce-dominated zone, and a spruce-with-deciduous-elements zone. Radiocarbon dates from the NAP-pine zone at Trolinger Spring are approximately 25,600 B.P. and 32,200 B.P., suggesting mid-Wisconsin interstadial age. The spruce-dominated zone may indicate the beginning of late-Wisconsin full glacial conditions. Radiocarbon dates from Boney Spring are approximately 13,700 B.P. and 16,580 B.P., thus giving an approximate time frame for the spruce-with-deciduous-elements zone (Mehring and others, 1970).

King (1973) subsequently examined Boney and Trolinger springs in addition to Kirby, Koch, and Jones springs, Hickory County (Fig. 1) and again recognized the same three pollen-assemblage zones. Radiocarbon dates indicate the presence of a NAP-pine zone before 40,000 B.P. that existed until approximately 23,000 B.P. and was deposited during the mid-Wisconsin interstade. Cyperaceae and *Pinus* make up at least 60% of the total pollen within this zone. Spruce is commonly found in quantities of less than 1%. Other arboreal pollen types include *Betula*, *Quercus*, and *Salix*. Nonarboreal pollen includes Gramineae, *Ambrosia*-type, and other Compositae. This zone is interpreted as a pine-parkland (King, 1973).

Directly above the NAP-pine zone is the spruce pollen zone, dominated by up to 92% spruce and also containing some *Pinus* and Cyperaceae pollen. This zone began with the onset of late-

Wisconsin full-glacial conditions. King (1973) interprets it as a boreal spruce forest that lasted until approximately 16,500 B.P.

A short hiatus separates the spruce zone from the spruce-with-deciduous-elements pollen zone, characterized by lower spruce percentages (less than 38%) and by an increase in thermophilous deciduous tree pollen. This zone is indicated by pollen found in pulp cavities of mastodon tusks recovered from Boney Spring. Spruce is still the dominant pollen type but *Pinus*, *Quercus*, *Salix*, *Alnus*, *Frazinus*, *Ulmus*, *Corylus*, and *Ostrya/Carpinus* also occur. Most of the NAP is composed of Cyperaceae, Gramineae, and various composites. This zone is interpreted as a spruce forest with deciduous trees, indicating slightly warmer climatic conditions during a late phase of Wisconsin full-glacial conditions (King, 1973).

McMillan and King (1974) reported finding a Holocene pollen sequence in sediments from Phillips Spring, Benton County. The spring sediments contained pollen spectra that indicate a change from arboreal pollen dominance, primarily *Quercus*, to nonarboreal pollen dominance. Radiocarbon dating places this change at 7800 B.P. Dominance of nonarboreal pollen continues until at least 4000 B.P. (McMillan and King, 1974).

King and Lindsay (1976) reiterated the previously mentioned studies (King, 1973) and related the findings to archaeological deposits at Rodgers Shelter, Benton County, and to megafauna assemblages associated with the pollen deposits. King and Lindsay (1976: 76, Table 4.2) summarized the radiocarbon dates, flora, and fauna from Trolinger and Boney springs (Table 1).

Old Field swamp, Stoddard County (Fig. 1) was cored and analyzed for pollen by King and Allen (1977). Two vegetation changes are indicated between 9000 and 3000 B.P. At approximately 8700 B.P., arboreal pollen composed largely of *Quercus*, *Frazinus*, and *Cephalanthus* was replaced with Gramineae and other NAP. A second change occurred at approximately 5000 B.P. when trees again began to increase. These changes are interpreted as reflecting the expansion and subsequent reduction of the Prairie Peninsula in southeastern Missouri between 8700 and 5000 B.P. (King and

TABLE 1. SUMMARY OF THE INFERRED DOMINANT VEGETATION AND FAUNAL ASSEMBLAGES FROM SPRING DEPOSITS IN THE WESTERN MISSOURI OZARKS

Inferred dominant vegetation	C ¹⁴ years B.P.	Megafaunal assemblages
oak-hickory forest	2,000	
(no data)	6,000	(no data)
	10,000	
spruce with deciduous trees	14,000	mastodon, tapir, ground sloth, deer, giant beaver, horse
	18,000	
spruce forest	22,000	(no data)
	26,000	
open pine parkland	30,000	mastodon, horse, muskox
	34,000	

From King, J.E., and Lindsay, E.H., 1976

Allen, 1977). Similar evidence for eastern expansion of the Prairie Peninsula has been found in pollen records from the northern midcontinent.

Watts and Bright (1968) record a shift to more prairie on the upland and a decrease in trees from a pollen sequence at Pickerel Lake, Day County, South Dakota between 8000 and 4000 B.P. At Kirchner Marsh, Dakota County, Minnesota, Winter (1962) also recorded an increase in nonarboreal pollen occurring between approximately 7200 and 5000 B.P. A migration of the prairie/forest ecotone of about 120 km northeastward occurred in western Minnesota between 8000 and 4000 B.P. (Wright, 1968). Brush (1967), Durkee (1971), and Van Zant (1976, 1979) record a shift from forest to prairie between 8000 and 3000 years ago in Iowa. In Illinois, E. Gruger (1972) recorded an undated rise in herb pollen and decrease in oak after the late Pleistocene spruce decline. Pollen records show that a mid-Holocene dry period lasting approximately 4000 years occurred between 8000 and 4000 years ago and reached maximum warm and dry conditions about 7000 B.P. (Webb and Bryson, 1972; Wright, 1971). At the same time the Prairie Peninsula reached its maximum eastward extent (Bernabo and Webb, 1977).

In northeastern Kansas, J. Gruger (1973) reported pollen spectra from Muscotah and Arrington marshes, Atchison County, indicating a forest grassland containing pine, spruce, and birch with alder and willow occurring locally, present between 25,000 and 23,000 years ago. Subsequently, the vegetation changed to a spruce forest that lasted until at least 15,000 B.P. As a result of a hiatus, no data from the marshes are available between 15,000 and 11,000 B.P. From approximately 11,000 to 9000 years ago a mixed deciduous forest and prairie occurred. At 9000 B.P., a dominance of prairie elements occurred in northeastern Kansas that lasted until approximately 5000 B.P. Development of prairie border vegetation then occurred (J. Gruger, 1973). J. Gruger (1973) also reports an *Ambrosia* rise in the youngest sediments and the presence of two grains of *Zea mays*, indicating settlement of the area.

Big Lake, Mississippi County, Arkansas contained a 160-cm pollen sequence, which has a radiocarbon basal date of less than 180 B.P., the approximate time of the New Madrid 1811-1812

earthquake (King, 1978). The pollen spectra from the core reflect vegetational change consistent with the modification of the landscape by humans since settlement of the area. The major feature of the pollen diagram is a decrease in oak and an increase in willow beginning between 70 and 80 cm, which may indicate the start of large lumbering operations in 1905 when a major drainage system was completed in the area. A second feature of the pollen sequence is a decrease or disappearance of several other trees and shrubs above 40 cm. In the top 40 cm of the sequence, only *Taxodium* and *Cephalanthus* appear to increase. A Chenopodiineae (Chenopodiaceae/Amaranthaceae) peak at 30 cm may indicate final drainage of the area when mudflats would have been created, opening the major habitat of *Amaranthus tuberculatus* for colonization. This possible source for the Chenopodiineae could have established itself quickly on the newly drained areas and produced large quantities of pollen until the water level reached a new lower equilibrium (King, 1978).

Missing from the diagram is a distinct *Ambrosia* rise (King, 1978), usually found in conjunction with land clearance in the central United States (Van Zant, 1976,1979; Huber, 1980, 1985; King, 1981). The absence of a distinct *Ambrosia* rise may result from the shielding effect of forests bordering Big Lake. Another possibility is that the *Ambrosia* rise may not be prominent in this area (King, 1978).

As part of his study in the western Missouri Ozarks, King (1973) analyzed six bryophytic polsters for modern pollen rain. Three samples were collected from the oak-hickory forests near Trolinger Spring, Hickory County and Boney Spring and Rodgers Shelter, Benton County. In Shannon County three moss polsters were collected from pine-oak forests at Round Spring State Park, Alley Spring State Park, and a native pine area 10 km north of Eminence, Missouri. The oak-hickory sites have up to 30% *Quercus* and high values of *Ambrosia*-type pollen (up to 40%) that may reflect abandoned farmland in the area (King, 1973). Pine values are low, less than 5%. *Fraxinus* values are approximately 15%. At the pine-oak sites, pine values reach 30%, oak 40%, and *Fraxinus* 15%. There is less NAP pollen at the pine-oak sites than at the oak-hickory sites (King, 1973).

Peterson (1978) analyzed 29 samples of surface sediments from lakes and ponds in Illinois, Missouri, and Kentucky for modern pollen rain. Seven of his sites occur in Missouri. Based on his data, Peterson (1978) presented isopoll maps for four pollen taxa, *Quercus*, *Ambrosia*, *Carya*, and *Ulmus*. The maps indicate values of approximately 30% for *Quercus* and *Ambrosia*, 3% for *Carya* and *Ulmus*. These values are calculated as a percent of the pollen sum, which excludes spores, aquatics, and unidentified grains (Peterson, 1978).

Pollen investigations of archaeological sites in the United States have yielded the best results in the arid southwest or in special circumstances where archaeological material occurs in a peat or marsh deposit (King, 1985). King (King, Klippel, and Duffield, 1975) reported that he was unable to extract pollen in quantities large enough to count from samples from Rodgers Shelter, Graham Cave, or the Old Fort Site, in Missouri, or Browsers Hole, a wet Pleistocene-age cave deposit in Illinois. However, Hall (1977a, 1977b, 1977c, 1979a, 1979b) and Henry, Butler, and Hall (1979) have been able to deduce Holocene environmental change from several archaeological sites and rock shelters in central and northeastern Oklahoma. Huber and Bailey (1983) identified *Zea mays* pollen from archaeological sediments from the Sand Lake Site, La Crosse, Wisconsin. King and Lindsay (1976), King (1985), Baker and Van Zant (1980), and others have been able to correlate occupational horizons from archaeological sites with stratigraphically continuous pollen sequences from the same region for several sites in the midwest. For an overview of palynological applications to archaeology, see King (1985).

Phytolith Studies

A phytolith is any microscopic mineral deposit formed within and between plant cells (Mulholland and Rapp, 1985). The term is restricted in this study to opaline silica ($\text{SiO}_2 \cdot n\text{H}_2\text{O}$) deposits. Phytolith type, form, and size can vary greatly within a single plant species (Rovner, 1983). Sizes range from approximately 1 to 1000 micrometers (Moody, 1972; Rovner, 1983). However, most phytolith studies have stressed the 5 to 50 micrometer range. Phytoliths are most abundant in the

Gramineae, Juncaceae, Cyperaceae, and Equisetaceae plant families. Although first investigated in the early 1800s, most detailed studies occur after the mid-1900s (Mulholland and Rapp, 1983; Mulholland, Rapp, Huber, and Selness, 1983).

Twiss, Suess, and Smith (1969) presented a morphological phytolith classification based on modern plant material. Their classification scheme consisted of four classes and 26 types of phytoliths. Three of the phytolith shape classes appeared to be correlative to three Gramineae subfamilies. The fourth class appeared to be diagnostic to Gramineae but not restricted to a certain subfamily. Twiss and others (1969) were then able to correlate phytoliths from different sediments to Gramineae subfamilies, thus setting the stage for paleoenvironmental reconstruction based on phytolith analysis.

Brown (1984) has developed an expanded phytolith key based on 112 grass species from central North America. The 112 species examined represented 52 genera, 16 tribes, and 4 subfamilies. Brown's (1984) key divides phytoliths into eight major classes, which are then subdivided based on minor shape characteristics. Mulholland and Rapp (1985) have devised a key based on 40 grass species that divides the phytoliths into ten shape categories. Their key is used in this study and will be discussed in more detail later. The investigations by Brown (1984) and Mulholland and Rapp (1985) both indicated that phytolith shapes were not as restricted to subfamilies as previously thought (Twiss and others, 1969).

Since the publication of Rovner's (1971) summary article, several phytolith studies have been undertaken. Moody (1972) compared phytolith and pollen from an archaeological site near Colstrip, Montana and was able to show a change in grass types based on phytolith data in conjunction with a decrease in *Pinus* and a rise in *Ambrosia* pollen. Bartoli and Guillet (1977) were able to establish the change from heather to pine at a location in France based on both phytolith and palynological data. Based on phytoliths, Schreve-Brinkman (1978) found a correlation between an increase in grass percentages and cold periods represented by pollen spectra for a 50,000-year period in the El Abra Corridor of the eastern Cordillera of Colombia, South America. Kurmann (1981) compared

phytoliths, pollen, and fungal spores from soils in Kansas. Her investigation demonstrated the need to use phytoliths in conjunction with pollen to differentiate shortgrass and tallgrass prairies where pollen identification is not specific enough to do so.

Several phytolith studies have centered on the identification of cultigens in archaeological sites, specifically corn (*Zea mays*). Pearsall (1978) was able to identify the presence of corn, based on the size of cross-shaped phytoliths, at an archaeological site dated at 2450 B.C. in coastal Ecuador. Piperno (1984) used both size and three-dimensional morphology of cross-shaped dumbbells to demonstrate the presence of corn at archaeological sites in the Pacific watershed of central Panama. Both Pearsall (1978) and Piperno (1984) investigated phytolith assemblages of local grasses and local varieties of corn to distinguish their corn phytolith size and shape criteria. However, Piperno (1984:381) warns:

“...each geographical region must be considered a separate problem. When embarking on phytolith studies, the wild grasses in each study area should be carefully evaluated for phytolith size and morphology. For instance, the present data should not be indiscriminantly extended to the American northeast or southwest, where species composition of mature grass cover is very different than that of Central America.”

Piperno's (1984) statement that phytolith criteria for the identification of corn may not apply in other parts of the Americas is further substantiated by S.C. Mulholland (1985, oral commun.), who is presently investigating Mandan corn varieties. She reported that phytoliths from Mandan corn grown in Minnesota have size ranges different than those reported by Pearsall (1978) and Piperno (1984).

Mulholland, Rapp, and Gifford (1982) have demonstrated the need to use computer analysis to identify multiple species from phytolith assemblages found in sediments. For an overview of phytolith applications, see Rovner (1983).

THE STUDY LOCALITY: ITS SETTING

The southeast Missouri Ozarks are located within the stable Mid-Continent area and have as their principal structural feature the Ozark uplift (McCracken, 1967) or Ozark dome (Bretz, 1965). It has been the focal point of mild repeated uplifts since Precambrian time (McCracken, 1967) and has been a land area continuously since Pennsylvanian time (Bretz, 1965). One of the oldest land areas in the United States, the Ozark dome has gone through several cycles of uplift and base-leveling since the Pennsylvanian (Bretz, 1965). The Ozark dome is also referred to as the Ozark Plateaus and Ozark province (Bretz, 1965; Fenneman, 1938).

The Ozark Plateaus physiographic province as defined by Fenneman (1938) is an area of 103,600 sq km that includes the relatively high country south of the Missouri River, east of the Prairie Plains of Kansas and Oklahoma, north of the Arkansas valley, and west of the Mississippi Valley and Embayment. The province also has an eastern extension called the Shawneetown Ridge, a linear structure extending across the Mississippi River into southern Illinois. Five subdivisions of the Ozark Plateaus province are recognized (Bretz, 1965; Fenneman, 1938) based on topographic, altitudinal, and lithologic differences. They are:

1. the Salem Platform or Plateau of southcentral Missouri and northcentral Arkansas;
2. the St. Francois Mountains, an island of crystalline rocks entirely surrounded by the Salem Plateau;
3. the Shawneetown Ridge in southern Illinois;
4. the Springfield Platform or Plateau of southwestern Missouri and northwestern Arkansas; and
5. the Boston Mountain Plateau in northern Arkansas and eastern Oklahoma.

The term "Ozark Plateau" is restricted by the Missouri Geological Survey to Fenneman's (1938) Salem subdivision, although all the above mentioned subdivisions are considered part of the "Ozark Province" according to Bretz (1965). The study area is located on the Salem Plateau (Fig. 2). It is the largest of the Ozark Province subdivisions and composes most of the summit area of the topographic dome (Bretz, 1965).

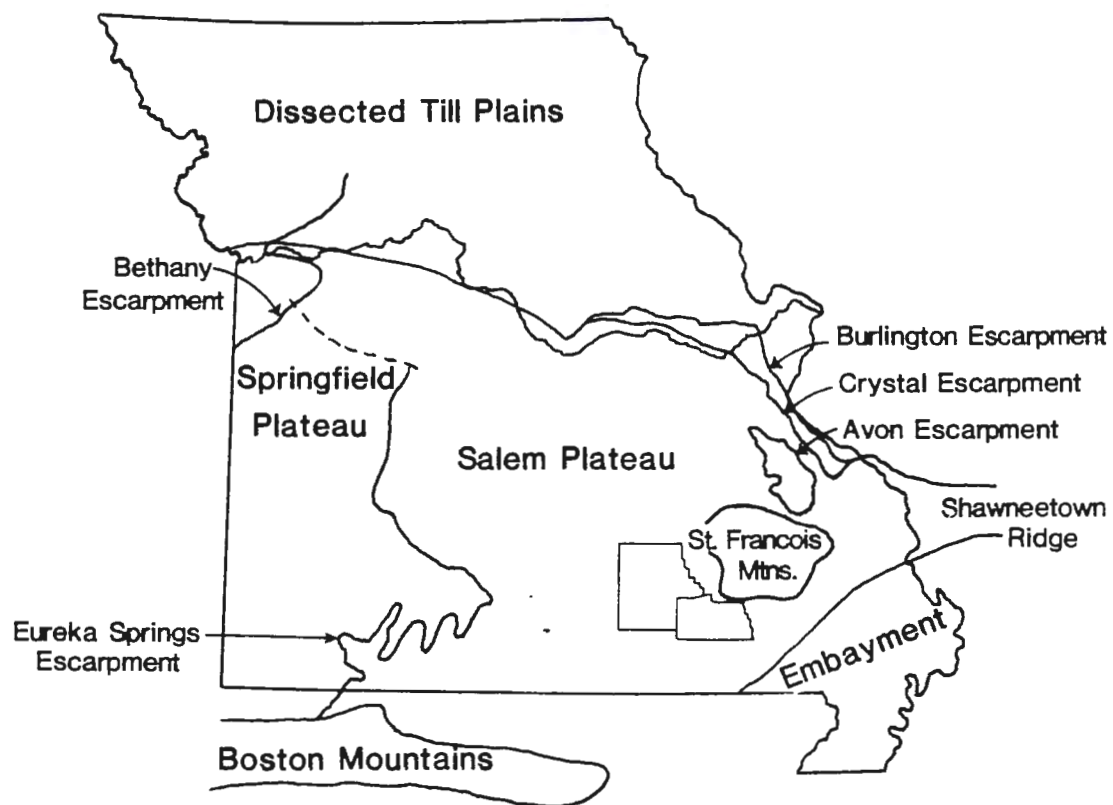


Figure 2. Subprovinces and escarpments of the Ozark Plateaus province. Shannon and Carter counties are outlined. Redrawn from Bretz, J. H., 1965.

Sauer (1920) subdivides the Ozark highlands into eight geographic provinces (Fig. 3). In his geographic scheme, the study area lies mostly in the Courtois Hills and the Central Plateau. He characterizes the Courtois Hills as a deeply dissected country with steep-sided hills and chert covered ridges. The Central Plateau, on the other hand, has not been extensively dissected and has a large number of small prairies divided by streams (Sauer, 1920).

Archaeological investigations, of which this study is a part, have centered on the Current River valley. Originating at Montauk Springs in the southcentral Ozarks, the Current River flows for 225 km to the Missouri-Arkansas border and then approximately another 50 km in Arkansas before entering the Black River. In Missouri, the Current River drains an area of 5490 sq km, draining a basin approximately 145 km in length and 80 km at its maximum width (Homyk and Jeffery, 1967).

Four springs of first magnitude feed the Current River; one of these is Big Spring, the largest single-outlet spring in the United States (Homyk and Jeffery, 1967; Jenkinson, 1973). These four springs together with other natural springs contribute approximately 60% of the Current River's flow (Harvey and Vineyard, 1967; Jenkinson, 1973), giving it the highest sustained base flow in the state (Homyk and Jeffery, 1967). In Carter and Ripley counties, the Current River valley is considered the boundary between the southeastern and southern slopes of the Salem Plateau (Bretz, 1965).

The study area lies approximately 550 km southeast of the maximum extent of Wisconsin glaciation, the Des Moines lobe in central Iowa. The last glaciation to enter Missouri was the Kansan, whose southernmost boundary correlates approximately with the Missouri River (Flint and others, 1959; Flint, 1971; Mehl, 1962) and occurred about 600,000 years ago (Matsch, 1976).

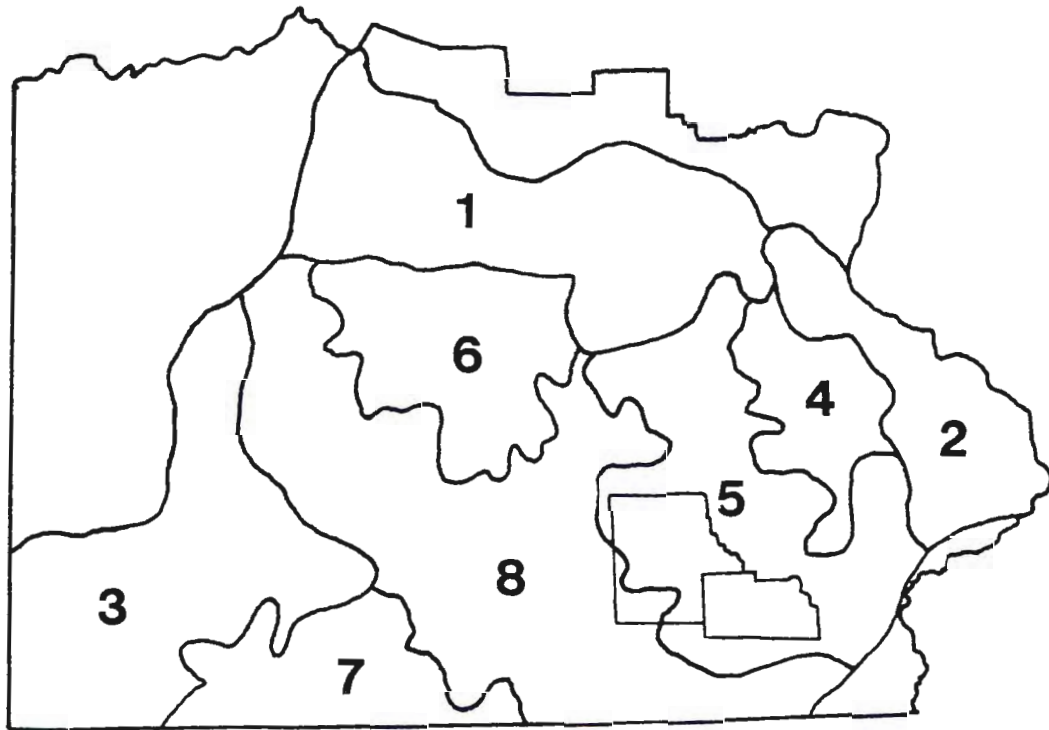


Figure 3. Geographical provinces of the Ozark Highland of Missouri. Shannon and Carter counties are outlined. Adapted from Sauer, C. O., 1920.

Explanation

1. Missouri River Border
2. Mississippi River Border
3. Springfield Plain
4. St. Francois Knob and Basin Region
5. Courtois Hills
6. Osage-Gasconade River Hills
7. White River Hills
8. Central Plateau

Bedrock Geology

Precambrian

Precambrian rock outcrops occur in both Shannon and Carter counties. They are part of the St. Francois Mountain Volcanic Supergroup (Anderson and others, 1979) and may be early-to-middle Precambrian in age (Hayes, 1967). Precambrian rocks occurring in the field area (Fig. 4) are primarily extrusive alkali rhyolite ash flow tuffs with minor trachytes (Anderson and others, 1979). The rocks are usually dark colored and porphyritic, showing well-developed flow structures (Hayes, 1961). Most of these were extruded as overlapping tuff sheets and lenticular flows, probably from several vents (Hayes, 1967). Stratigraphically, the felsitic flows are divided into two units (Hayes, 1961, 1967) separated in many places by a water-laid tuff (Hayes, 1961). The lower unit is characterized by a high potash-soda ratio (Hayes, 1961, 1967), whereas the upper unit had more normal ratios (Hayes, 1967).

Cambrian

Strata of Cambrian rocks lie unconformably on the Precambrian (Hayes and Knight, 1961; Howe, Anderson, and McCracken, 1967) and are considered to be of Late Cambrian age (Hayes and Knight, 1961). Only the two uppermost formations of the Upper Cambrian Series occur in the study area: the Potosi and Eminence formations (Anderson and others, 1979).

The older Potosi Formation (Fig. 4) is a massive, thickly bedded, fine-to-medium grained dolomite (Hayes and Knight, 1961) that contains abundant quartz druse (Hayes and Knight, 1961; Howe and others, 1967) associated with chert. Rocks of the Potosi Formation are usually brownish gray in color, weathering to a light gray (Hayes and Knight, 1961). When broken, the rock gives off a distinctive bituminous odor (Hayes and Knight, 1961; Howe and others, 1967). The Potosi ranges from 23 to 91 m in thickness with an average thickness of 61 m (Hayes and Knight, 1961).

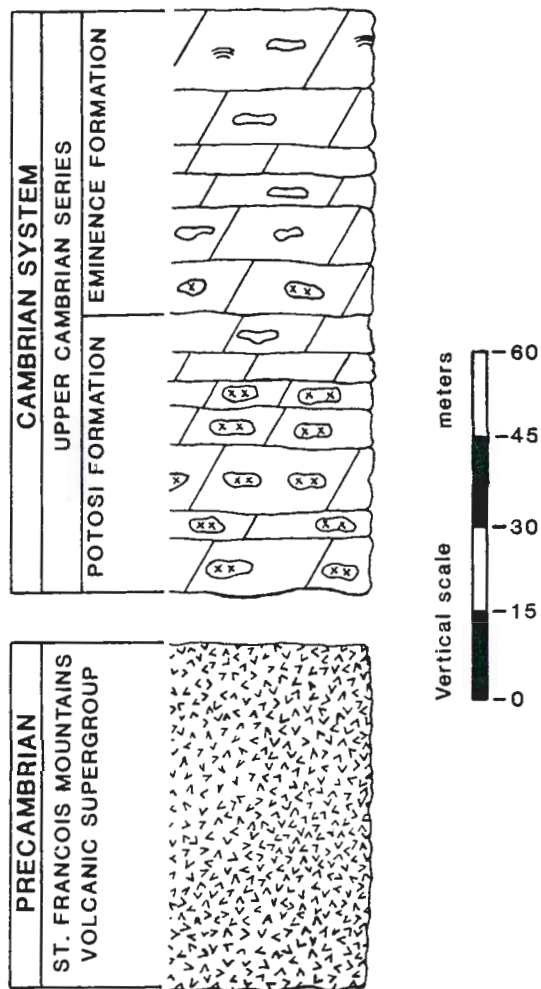


Figure 4. Precambrian and Cambrian stratigraphy of Shannon and Carter counties. Adapted from Anderson, K. H., and others, 1979 and Hayes, W. C., and Knight, R. D., 1961.

LEGEND



Dolomite



Cryptozoans



Dolomite containing nodules and beds of chert



Dolomite containing cavities lined with quartz druse



Chiefly alkali rhyolitic ash-flow tuffs with minor trachytes

Conformably overlying the Potosi is the Eminence Formation (Fig. 4), which is predominantly a medium-to-massive bedded, light gray, medium-to-coarse grained dolomite. Its thickness is approximately 60 to 75 m; in the upper half of the formation, a few small nodules and angular fragments of chert are found. The Eminence also contains small amounts of quartz druse similar to that in the underlying Potosi Formation. Chert boulders and blocks as large as two meters in diameter are found in some areas of the formation. In localized areas, white oolitic chert that often contains gastropod casts and molds may be found in the upper part of the formation. In some places, *Cryptozoon* masses occur near the top. Round Spring and other large springs, as well as several large caves, are found in the Eminence Formation (Hayes and Knight, 1961).

Ordovician

According to Martin, Knight, and Hayes (1961) and Anderson and others (1979), rocks of Ordovician age unconformably overlie the Cambrian. On the other hand, Howe, Anderson, and McCracken (1967:16) state:

“...sedimentation continued either unbroken or with only minor unconformity into Early Ordovician (Canadian) time.”

In any case, rocks of the Ozark dome dip radially (Martin and others, 1961; Howe and others, 1967; McCracken, 1967). Three Ordovician Series are found in Missouri (the Canadian, Champlainian, and Cincinnati) but only the Canadian Series occurs in the study area (Anderson and others, 1979). The Canadian Series is composed of the Gasconade, Roubidoux, Jefferson City, Cotter, Powell, and Smithville formations (Martin and others, 1961; Anderson and others 1979).

The Gasconade Formation (Fig. 5) is composed predominantly of a light-brownish gray cherty (Heller, 1954; Martin and others, 1961), fine-to-coarse grained, medium-to-massive bedded non-sandy dolomite (Heller, 1954). In the lowermost part of the formation is a sandstone unit designated the Gunter Member (Fig. 5) and composed of a medium-grained quartzose sandstone. In some areas, a basal conglomerate containing pebbles from the underlying Eminence dolomite has been found.

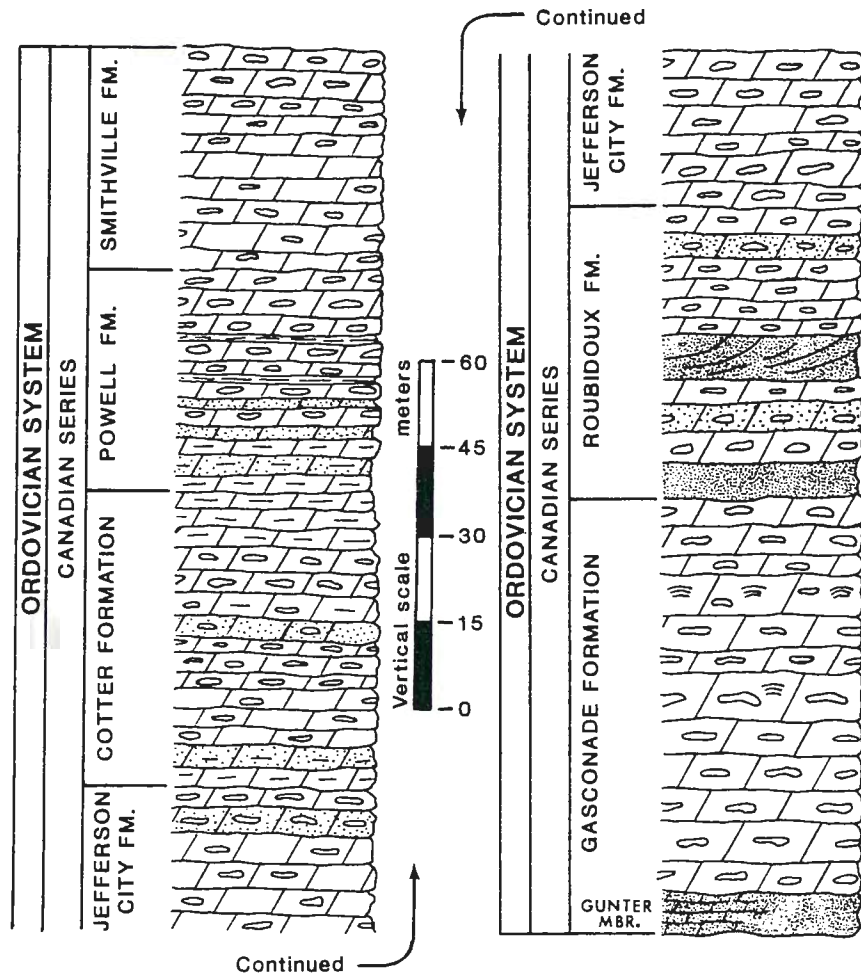
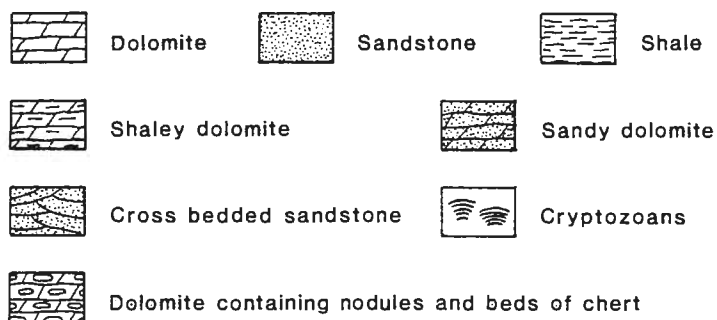


Figure 5. Ordovician stratigraphy of Shannon and Carter counties. Adapted from Martin, J. A., Knight, R. D., and Hayes, W. C., 1961.

LEGEND



The rest of the Gasconade Formation overlying the Gunter Member is divided into two parts. The lower part is a coarse crystalline dolomite characterized by an abundance of chert that often exceeds 50 percent of the total volume of rock. The upper part, however, is mainly a fine crystalline dolomite containing a relatively smaller amount of chert (Martin and others, 1961), rarely exceeding 10 percent by volume (Heller, 1954).

Variations in the composition of the chert characterize different parts of the Gasconade Formation. In the lowermost dolomite, just above the Gunter, the chert is often oolitic; some of the ooliths are bean-shaped and free. In the middle of the dolomite is a smooth, white, porcelaneous chert and a second type with a "dead" appearance. Small amounts of brown-and gray-banded chert are found in the lower half of the upper 7.5 to 9 m of the Gasconade Formation (Martin and others, 1961).

There are relatively few fossils found in the formation except for mollusks that commonly occur in the chert (Martin and others, 1961), although *Cryptozoon* masses are widely spread throughout (Heller, 1954; Martin and others, 1961). The most abundant of these are found 15 to 21 m from the top of the formation (Heller, 1954; Martin and others, 1961). The *Cryptozoon* are commonly found in the form of chert beds (Heller, 1954).

In the Ozark region, the Gasconade has an average thickness of 90 m that forms many of the almost vertical cliffs and bluffs in the central Ozarks. Caves and springs are also common in this formation (Martin and others, 1961).

Turner (1954) reported an Indian quarry in Laclede County where a "very fine grained white quartzite" was sought and stated that the quartzite bed was in the Gasconade Formation. Thus, the chert or the "quartzite" from the Gasconade Formation probably provided a source for aboriginal stone tool manufacture.

Overlying the Gasconade is the Roubidoux Formation (Fig. 5), consisting of sandstone, dolomitic sandstone, and cherty dolomite (Heller, 1954; Martin and others, 1961). It is predominantly a quartzose sandstone in central Missouri. In other areas of the state, the formation may contain as little as 10 percent sandstone with cherty dolomite composing the remainder. The

quartz sand is fine-to-medium grained, subrounded, and frosted. The weathered surfaces are gray and brown, whereas fresh sandstone is usually yellow, tan, or red at the surface and white in the subsurface. The dolomite component of the Roubidoux is finely crystalline, light gray to brown, and thinly to thickly bedded. Brown-to-gray banded oolitic, sandy chert can be found in individual beds (Martin and others, 1961).

The Roubidoux usually contains few fossils, although some of the chert contains localized fossiliferous deposits of mollusks. In many places the sandstone is characterized by well preserved ripple marks, mud cracks, and crossbedding. The formation ranges from 30 to 75 m in thickness, reaching its greatest thickness in the southwestern part of the Ozarks (Martin and others, 1961).

Above the Roubidoux is the Jefferson City Formation (Fig. 5), composed of light brown, medium-to-finely crystalline dolomite and argillaceous dolomite. Localized lenses of orthoquartzite, conglomerate, and shale occur in the formation. The Jefferson City Formation varies in stratigraphic succession from one locality to the next and is characterized by a finely crystalline, argillaceous dolomite called "cotton rock." In the Ozark region, another rock type informally known as "Quarry Ledge" occurs at approximately 10 to 12 m above the base of the formation. It is a thickly bedded, massive, brown, medium crystalline dolomite that weathers with a coarsely pitted surface (Martin and others, 1961).

The Jefferson City Formation occurs in outcrops around the perimeter of the Ozarks and is recognized in subsurface by its characteristic oolitic chert. Within the formation are zones of insoluble residue containing siliceous spicules commonly referred to as "spines." The average thickness of the Jefferson City Formation is 60 m, although it ranges from 40 to 105 m (Martin and others, 1961).

Frost-fractured and weathered chert occurs on the surfaces of hills and slopes and in stream beds associated with the Jefferson City Formation. Buried in hillside residuum just below the frost line, large round-to-oblong nodules or small boulders can be found that provided a good source of chert for the manufacture of stone tools. No known aboriginal quarrying operations have been found in connection with the Jefferson City Formation, but this is probably the result of the

widespread occurrence of its cherts and their proximity to the surface (McMillan, 1976a). The previously mentioned “cotton rock” occurring in the Jefferson City Formation can also be found on hillsides and in streambeds. It is easily worked and is also a useful resource for aboriginal tool manufacture (McMillan, 1976a).

Overlying the Jefferson City is the Cotter Formation (Fig. 5), composed for the most part of light gray to light brown, medium-to-fine crystalline, cherty dolomite. The formation is usually medium-to-thin bedded, containing thin interstratified beds of green shale and sandstone (Martin and others, 1961).

The Cotter Formation is composed of three parts. The lower part contains echinoderm fragments and is relatively free of chert. Oolitic chert and large siliceous oololiths characterize the middle part. The upper part is shaley and contains small quartz masses and brown quartzose oolitic chert. The average thickness of the Cotter Formation is 60 m, with a maximum thickness of 140 m (Martin and others, 1961).

Above the Cotter is the Powell Formation (Fig. 5), composed of medium-to-finely crystalline dolomite, and thin beds of green shale and fine-grained sandstone. The Powell averages 45 to 55 m in thickness (Martin and others, 1961).

The Smithville Formation (Fig. 5) is situated above the Powell. It is composed of dolomite containing a small amount of chert, and is characterized by Bryozoan fossils. The Smithville ranges up to 45 m in thickness and is distinguished from the similar underlying Powell by the characteristics of its insoluble residue (Martin and others, 1961).

Mississippian

An unconformity occurs after the Ordovician, so that rocks of Silurian and Devonian age are absent in the study area. Rocks of Mississippian age overlie Ordovician deposits in small areas in Shannon County; those that do occur in the study area are part of the Osagean and Chesterian series (Anderson and others, 1979).

The Osagean series is composed of the Pierson, Fern Glen, Reed Spring, Grand Falls, Burlington, and Keokuk formations (Spreng, 1961). However, only the Fern Glen, Burlington, and Keokuk formations occur locally (Anderson and others, 1979). The Osagean series is composed of fossiliferous limestones that are crinoidal, very cherty, and generally coarsely crystalline (Spreng, 1961).

The Fern Glen Formation (Fig. 6) is composed of gray, grayish-green, and red limestone, and red calcareous shale. In most areas, the lower portion is noncherty and the upper portion contains small nodules and layers of grayish-green to gray chert. At most localities the Fern Glen consists of three lithologic types: the lower, a noncherty, brown, thickly bedded, crinoidal limestone 1 to 3 m thick, containing a few quartz geodes in some areas; the middle, a red and/or green fossiliferous, calcareous shale approximately 3 to 6 m thick; and an upper, nodular cherty limestone 4 to 9 m thick that contains some quartz geodes. The formation ranges from 6 to 14 m in total thickness. The upper crinoidal limestone indicates that the Fern Glen Formation may be transitional with the overlying Burlington (Spreng, 1961).

The formation contains numerous fossils including brachiopods, corals, and crinoids. Commonly found fossils include the bryozoan (*Evactinopora sexradiata*), the brachiopods (*Spirifer vernonensis*, *S. rowleyi*, *Athyris lamellosa*, and *Cleiothyridina*), and the coral (*Cyathaxonia arcuatas*). Several species are restricted to this formation (Spreng, 1961).

Conformably overlying the Fern Glen is the Burlington Formation (Fig. 6), which is widespread and of uniform lithology in Missouri. It is composed of white to light buff, very coarsely crystalline, fossiliferous, crinoidal limestone. Layers of chert nodules are commonly found, particularly in the upper part (Spreng, 1961). Estimating the thickness of the Burlington has been difficult because the boundary with the overlying Keokuk Formation is obscure. However, it is believed that the Burlington Formation has a fairly uniform thickness throughout the state, seldom exceeding 30 m (Spreng, 1961).

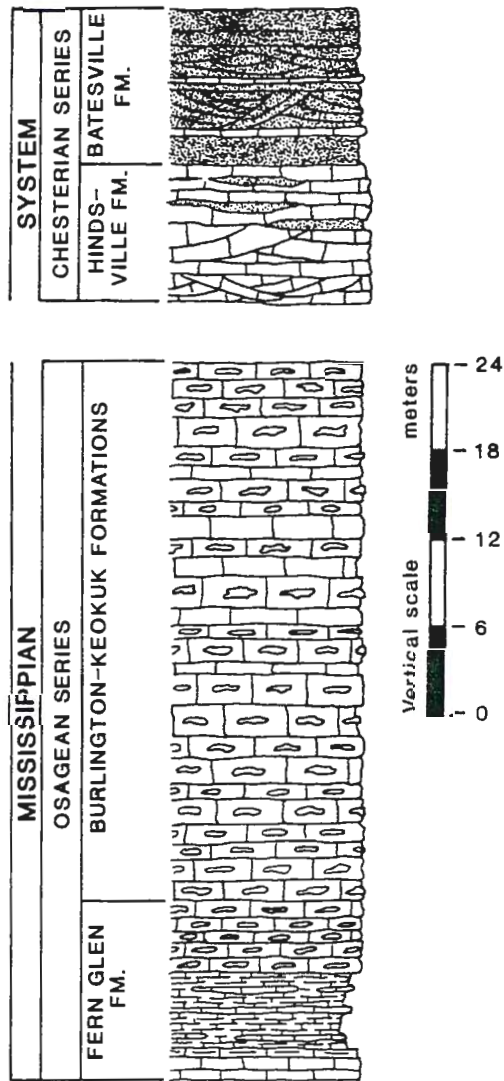
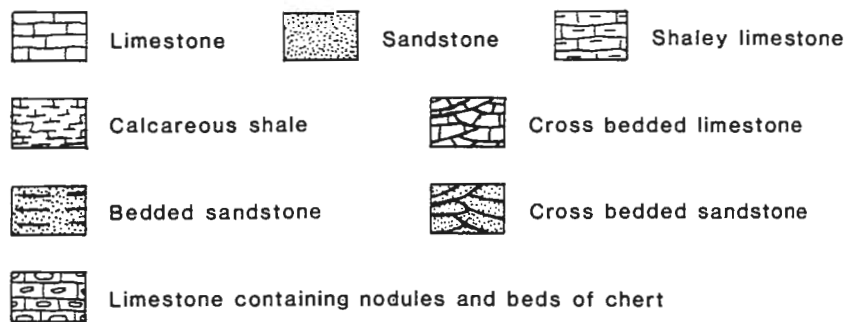


Figure 6. Mississippian stratigraphy of Shannon and Carter counties. Adapted from Spreng, A. C., 1961.

LEGEND



In Polk County, Missouri, extensive aboriginal quarries have been found above the Burlington Formation in residuum that are still visible today (McMillan, 1976a; Turner, 1954). Thus, the Burlington Formation also provided a chert source for stone tool manufacture.

The Keokuk Formation (Fig. 6) conformably overlies the Burlington in the study area, and like the Burlington Formation is widespread and has a uniform lithology. The formation is composed of a bluish-gray, medium-to-coarsely crystalline, medium-bedded limestone containing a large amount of light gray chert in the form of layers and nodules (Spreng, 1961).

In the Keokuk Formation (Fig. 6), the chert is irregularly distributed but appears to be more concentrated in the upper and lower portions. The chert is light gray and dense and has tripolic borders. It weathers to buff and reddish brown (Spreng, 1961).

Commonly found fossils occurring in the Keokuk limestone are brachiopods, *Buxtonia*, *Dictyoclostus*, *Linoproductus*, and *Marginirugus*. Other species of brachiopods include *Orthotetes keokuk*, *Cleiothyridina obmaxima*, *Echinoconchus alternatus*, *Spirifer logani*, and *Tetracamera* spp. Horn corals and bryozoans are found in the Keokuk, especially the bryozoan genus *Archimedes*. In the study area, the thickness of the Keokuk is approximately 15 m (Spreng, 1961).

Unconformably overlying the Keokuk Formation is the Chesterian Series. In the study area, this includes only the Hindsville and Batesville formations. In southwestern Missouri, the Cartersville lies at the bottom of the Series and the Fayetteville at the top (Spreng, 1961).

The Hindsville Formation (Fig. 6) is a light-to-dark gray, medium-to-finely crystalline, oolitic limestone. In some areas the limestone is interbedded with light gray, calcareous shale and siltstone. Cross-beds are commonly found in the limestone. In the upper part of the formation, sandstone lenses occur that are indistinguishable from the Batesville Formation. Glauconite is present in some places, giving the rock a greenish tinge (Spreng, 1961).

The contact of the Hindsville with the Keokuk Formation is irregular and marked by chert-pebble conglomerates that contain fish teeth. The Hindsville Formation ranges to 15 m in thickness (Spreng, 1961).

Overlying the Hindsville is the Batesville Formation (Fig. 6), a yellowish-brown, finely crystalline, calcareous sandstone containing discontinuous beds of thin gray, medium crystalline, oolitic limestone. Brachiopods and pelecypods are the most abundant fossils found in the formation, which ranges from 10 to 15 m in thickness (Spreng, 1961).

Soils

The soils of the study area were formed under forests and contain low quantities of organic matter and plant nutrients. The Ozark soils can be differentiated from all others in the state by their light color (Krusekopf, 1962) and were derived from cherty limestone, dolomite, and sandstone (Scrivner, Baker, and Miller, 1966). They belong to the Gray-Brown Podzolic and Red-Yellow Podzolic soil groups found in the southern portion of the North Central Region of the United States (Krusekopf, 1960).

The Clarksville-Fullerton-Talbott soil association is found in most of the study area, with the Lebanon-Nixa-Clarksville and Hobson-Clarksville soil association occurring in western Shannon and eastern Carter counties. The Lebanon-Nixa-Clarksville and Hobson-Clarksville soil association is composed of highly weathered, forested soils that occupy gently rolling to nearly level topography interspersed with steeply sloping areas bordering streams and drainages. The flat ridgetops show evidence of a thin loess cap less than a meter thick. The Clarksville-Fullerton-Talbott soil association is forested limestone-derived soils that compose a large part of the steeply sloping areas of the Current, White, Meramec, Osage, and Gasconade rivers' drainage basins. The main differences between these two soil associations are the chert content and depth to red high-clay subsoils (Scrivner and others, 1966).

Lebanon soils are found on ridgetops, are moderately well drained, and are relatively free of chert in the upper meter. At depths of 75 to 82 cm, Lebanon soils have a dense impermeable fragipan layer underlain by a cherty red clay. Tree root penetration is limited by the fragipan;

therefore, only scrub oak timber occurs under natural conditions. In late summer, Lebanon soils are droughty as a result of the low water-holding capacity of underlying materials (Scrivner and others, 1966).

Hobson soils are found on topographies similar to Lebanon soils. They also have a fragipan, but are developed from sandstone, and are even more droughty than Lebanon soils (Scrivner and others, 1966). Nixa soils are found on more sloping topography. The fragipan is less distinct and the soils have cherty fragments above the fragipan (Scrivner and others, 1966).

Found on steeper slopes, Clarksville soils are very cherty in the upper 72 cm and have red clay subsoils that contain less chert than the 72 surface centimeters. They are also the most droughty (Scrivner and others, 1966).

Talbott soils contain very little chert. Their red clay subsoils usually occur at less than 25 cm (Scrivner and others, 1966).

All of these soils are low in fertility and most are forested, with low-to-moderate moisture capacity (Scrivner and others, 1966). The map (Fig. 7) shows the general distribution of Missouri's major soil series and their association.

Climate

As an inland state, Missouri has an essentially continental climate (Homyk, Harvey, and Jeffery, 1967; McQuigg, 1969). Weather changes occur frequently, both from day to day and from season to season. Missouri receives cold air moving down from Canada, warm moist air from the Gulf of Mexico, and dry air from the west (McQuigg, 1969).

The inland location of Missouri causes cold winters and hot summers, although long periods of either very cold or very hot temperatures are unusual. In winter, occasional periods of mild above-freezing temperatures occur. During the hot summer season, periods of cool, dry weather occasionally break up the periods of hot, humid weather (McQuigg, 1969).

Figure 7. Distribution of the major soil areas of Missouri. Shannon and Carter counties are outlined. Redrawn from Scrivner, C.L., and Miller, B.S., 1966.

Explanation

NORTHERN MISSOURI LOESS AND LOESS-TILL LANDSCAPES

Prairie and Prairie-Forest Transition Natural Vegetation

M = Marshall-Knox	AG = Adair-Shelby-Grundy-Lagonda
SA = Sharpsburg-Grundy-Adair-Shelby	AS = Adair-Shelby-Seymour-Edina
SG = Sharpsburg-Grundy-Lagonda-Pershing	and Armstrong-Gara-Pershing
G = Grundy-Pershing	PM = Putnam-Mexico

Forest Natural Vegetation

MW = Menfro-Winfield-Weldon	LK = Lindley-Keswick-Hatton
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SOUTHERN MISSOURI RESIDUAL AND LOESS-RESIDUAL LANDSCAPES

Prairie and Prairie-Forest Transition Natural Vegetation

SD = Summit-Newtonia-Parson-Dennis	GC = Gerald-Craig-Eldon and
PD = Parsons-Dennis-Bates	Newtonia-Baxter

Forest Natural Vegetation

B = Bolivar-Mandeville	CF = Clarksville-Fullerton-Talbott
BB = Baxter-Bodine	AH = Ashe-Tilsit-Hagerstown
GB = Gasconage-Bodine-Clarksville	HT = Hagerstown-Tilsit
LC = Lebanon-Nixa-Clarksville and	UF = Union-Fullerton-McGirk
Hobson-Clarksville	ML = Memphis-Loring

ALLUVIAL VALLEY LANDSCAPES

Missouri and Upper Mississippi River

S = Sarpy-Haynie-Onawa-Wabash

Southeastern Missouri

C = Commerce-Hayti-Caruthersville	SF = Sharkey-Alligator-Forrestdale
DD = Dexter-Dubbs-Dundee-Baskett	WC = Waverly-Calhoun

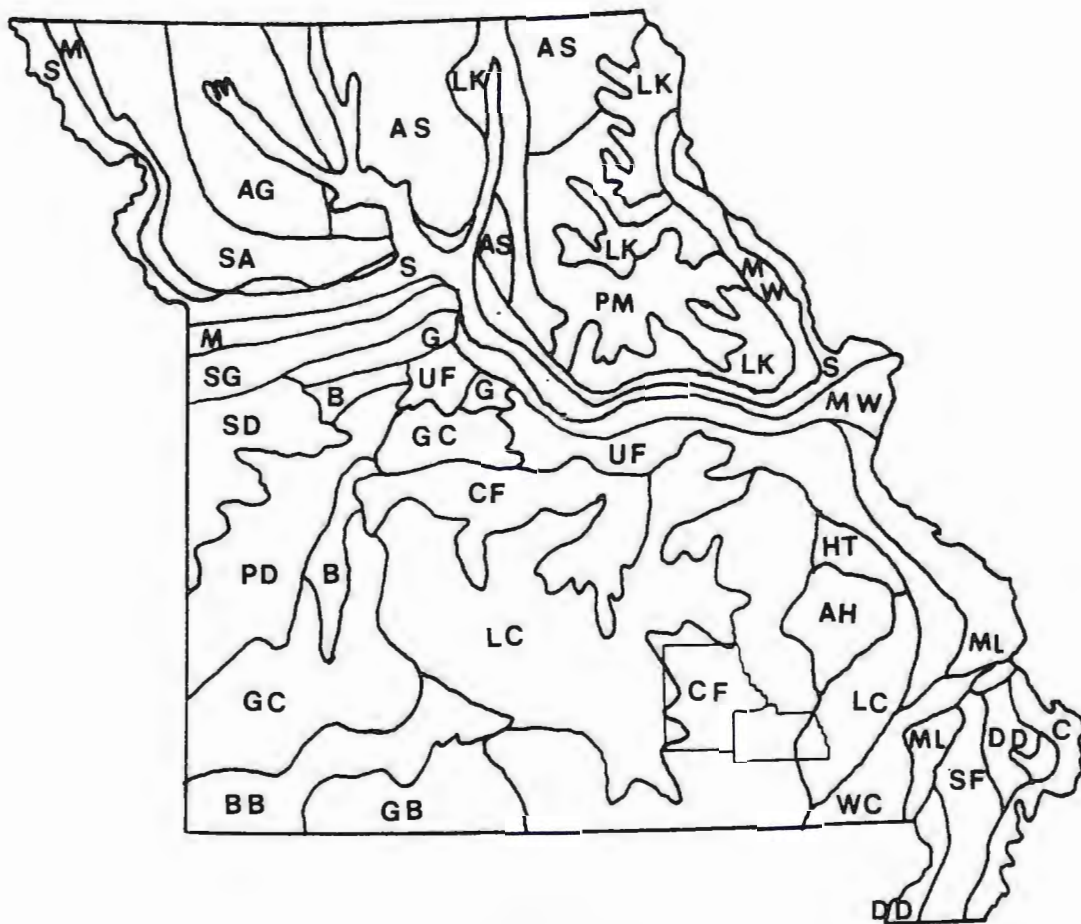


Figure 7.

In attempting to describe the climate of southeast central Missouri, it is convenient to speak in terms of averages and means. This is a useful way to indicate the average weather during different seasons but does not adequately express the complexity of climate (McMillan, 1976a). In any attempt to characterize contemporary weather patterns one must note Transeau's warning (1935:436) that:

"... the extremes of the factors are vastly more important than the means ... an extreme drought marked by lower precipitation, higher evaporation, higher temperatures, and more intense light can change vegetation more in a few years than a century of favorable weather conditions."

Table 2 shows contemporary climatic data for the study area.

Approximately 42% of Missouri's precipitation occurs from May through August, during which time the rainfall frequently occurs as severe thunderstorms. More than 25 cm of rainfall has been recorded in 24 hours (Homyk and others, 1967). Evaporation averages approximately 91.5 cm over the state annually (Homyk and others, 1967).

The study area lies near the border of Borchert's (1950:29) Climatic Region 4 (the Prairie Peninsula) and Climatic Region 2 (the Southeast and Eastern Seaboard) (Fig. 8). The major characteristics of the weather patterns described by Borchert (1950:29) for Region 4 are:

1. low winter rainfall and snowfall;
2. occasional major summer droughts with a tendency for major summer droughts to occur synchronously within the region; and
3. the continental source and trajectory of the mean airstream that blankets the region during dry periods.

Borchert (1950:32) described the major characteristics of Climatic Region 2 as follows:

1. rainy winters;
2. the region receives as much or somewhat more rain in summer than in winter, but the summers are sunnier; and
3. winter, because of its high rainfall, is the season during which the region is distinctly different climatically from the rest of eastern America.

Borchert (1950:34) also states that:

"...the climatic region coincident with the prairies (Region 4) is actually a broad boundary zone between steppe and forest" (Fig. 9).

TABLE 2. CONTEMPORARY CLIMATIC DATA FOR THE STUDY AREA

TEMPERATURE (degrees Celsius)*

Average Monthly Temperatures for the Period 1931-1960

Jan.	Feb.	Mar.	April	May	June	July	Aug.	Sept.	Oct.	Nov.	Dec.
1.9	3.7	7.8	14.0	18.3	23.3	25.6	25.1	21.0	15.1	7.8	3.2

Mean Annual Temperature 13.9

January mean maximum temperature 8.4

January mean minimum temperature 3.3

July mean maximum temperature 32.8

July mean minimum temperature 18.9

PRECIPITATION (centimeters)*

Mean Annual Precipitation 112.22

Spring 34.90

Summer 27.56

Fall 26.39

Winter 23.37

GROWING SEASON

Average date for last killing frost April 20 March 29*

Average date for first killing frost October 250 November 2*

Average number of frost-free days 205 213

* Adapted from McQuigg (1969)

@ Adapted from Decker (1955)

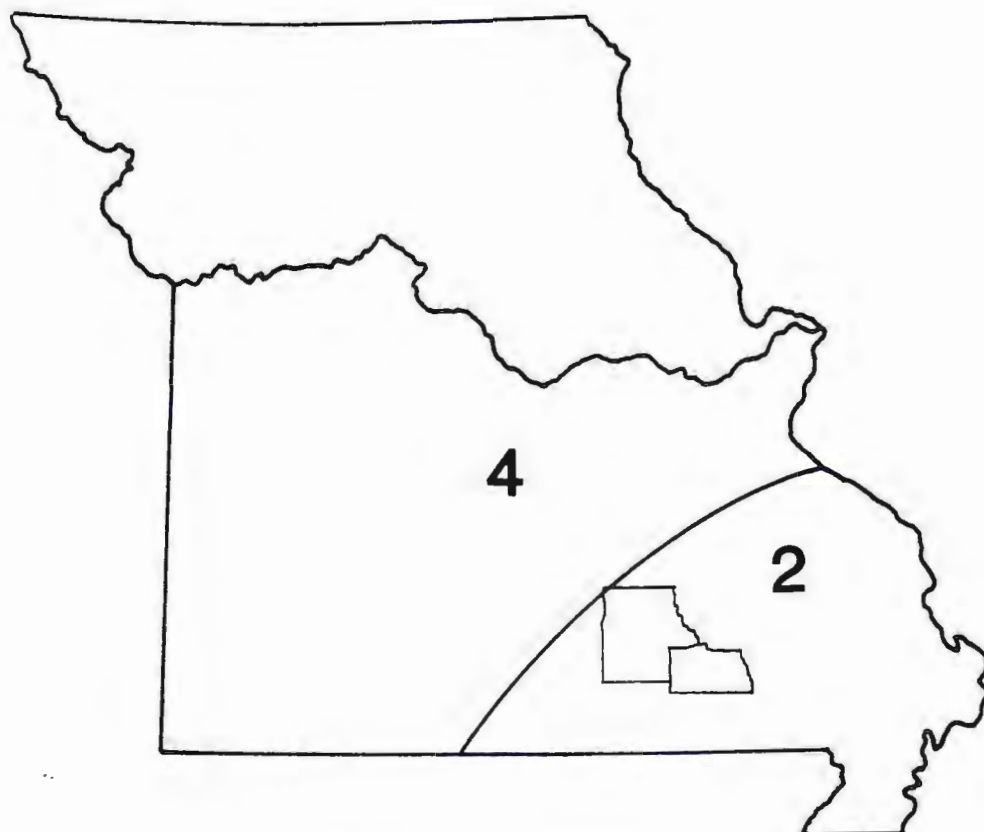


Figure 8. Border of Borchert's Climatic Regions
2 and 4. Shannon and Carter counties are outlined.
Adapted from Borchert, J. R., 1950.

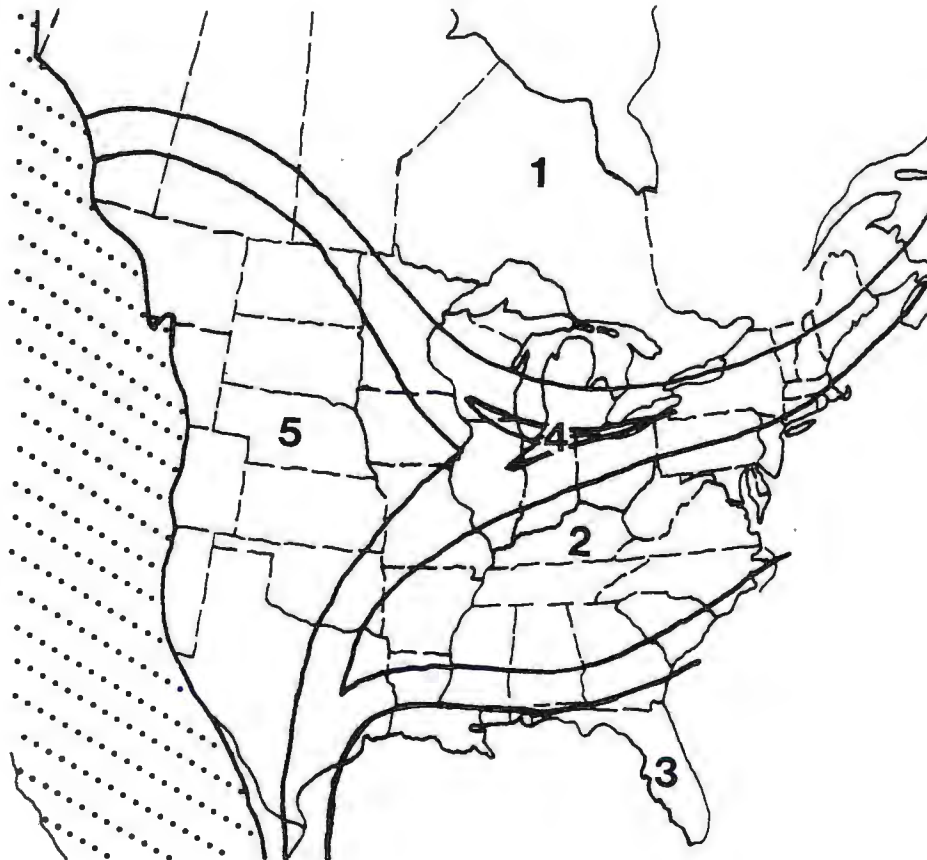


Figure 9. Climatic regions of eastern America.
Redrawn from Borchert, J. R., 1950.

Explanation

1. Northeast
2. Southeast and Eastern Seaboard
3. South Atlantic and Gulf Coasts
4. Prairies
5. Western Great Plains

Thus, the weather patterns described for Region 4 often produce droughts that have great ecological impact on ecotones. Because droughts usually favor grasses over arboreal species, the prairie-forest border may change, with grasslands increasing and decreasing in relation to a fluctuating climate (McMillan, 1976a). Therefore, the study locality lies in a kinetic area of the prairie-forest boundary.

Flora

Floristically, the study locality is located in Steyermark's (1963) Ozark Region (Fig. 10) and more specifically in K  chler's (1964) oak-hickory-pine forest (Fig. 11). The floral regions of Missouri are similar to the physiographic areas previously described, but the boundaries are not quite the same. Characterized by a diversified flora, the Missouri Ozark Region contains the greatest number of species of any part of the state (Steyermark, 1963). Many microclimates and microenvironments occur within this area, each possessing a characteristic assemblage of plants (Huber and Rapp, 1981; revised 1983).

The Ozark Region forest flora is pine-oak and oak-hickory woodlands. Although the Ozark forests are considered to be oak-hickory-pine forest (K  chler, 1964) or pine-oak and oak-hickory (Steyermark, 1963), hickory is a relatively minor element (King, 1973). The forest flora with its herbaceous components belong to a Carolinian flora (Dice, 1943; Steyermark, 1963) with a slight dominance of southern species, floristically intermediate between austral and boreal phases. The upland herbaceous species are usually plants that range from the Appalachian plateau to the grassy plains (Steyermark, 1963).

Lowbush blueberry (*Vaccinium vacillans*) is the dominant understory on acid soils. Limestone-derived soils have floras consisting of southern buckthorn (*Blumelia* spp.), deciduous holly (*Ilex decidua*), linden (*Tilia americana*), walnut (*Juglans nigra*), pawpaw (*Asimina triloba*), chinquapin oak (*Quercus prinoides* var. *acuminata*), blue ash (*Fraxinus quadrangulata*), sugar maple (*Acer saccharum*), and other forest species (Steyermark, 1963).

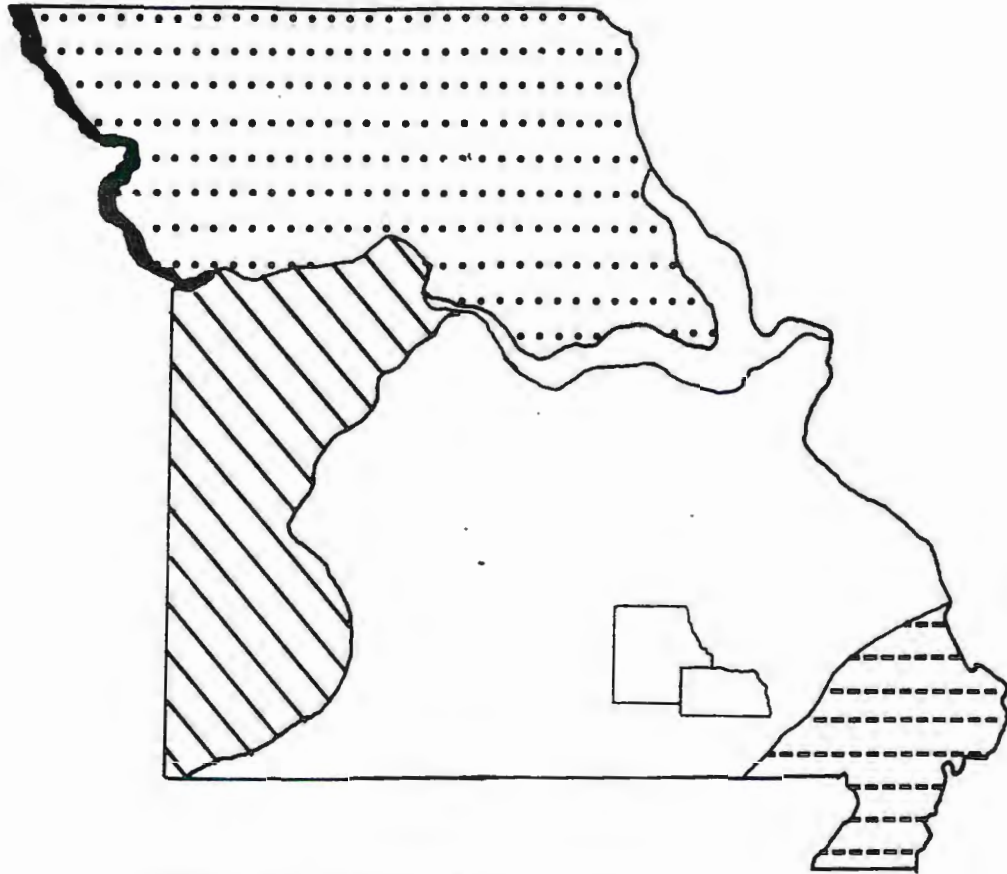


Figure 10. Principal plant regions of Missouri.
Redrawn from Steyermark, J. A., 1963.

Explanation

	Prairie Region (Loess mounds)
	Prairie Region (unglaciaded)
	Prairie Region (glaciaded)
	Ozark Region
	Southeastern Lowlands

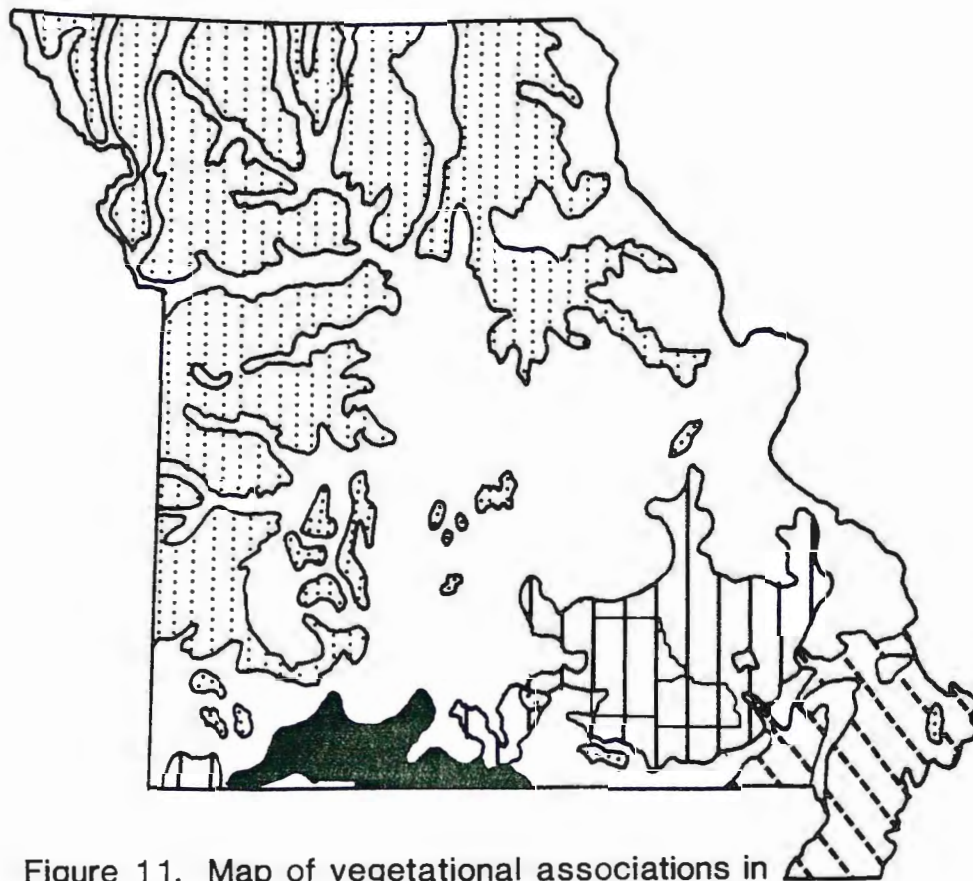







Figure 11. Map of vegetational associations in Missouri. Shannon and Carter counties are outlined.
Adapted from Küchler, A. W., 1964.

Explanation

-  Oak-hickory-pine forest (*Quercus-Carya-Pinus*)
-  Southern floodplain forest (*Quercus-Nyssa-Taxodium*)
-  Cedar glades (*Juniperus-Quercus-Sporobolus*)
-  Mosaic of Oak-hickory forest (*Quercus-Carya*) and Bluestem prairie (*Andropogon-Panicum-Sorghastrum*)
-  Oak-hickory forest (*Quercus-Carya*)

Several herbaceous plants have their closest associations with southern coastal plain species. Other species are usually found in a more northern boreal habitat and represent relict species that have survived in the Ozarks since the retreat of the Laurentide ice sheet. Many plants that have rare and isolated occurrences are at or near the limits of their geographic range (Steyermark, 1963).

The Ozark region has never been glaciated and thus has been open for migration since Tertiary times. Some species are restricted to the eastern, western, or southern edges of the region whereas others are widespread throughout. The southeastern Ozark Region is characterized by flora with an Alleghenian relationship. Many southwestern, southern, or western species are found on exposed south- and west-facing slopes. In the deeply eroded V-shaped valleys and north-facing bluffs, many of the more northern ranging species occur (Steyermark, 1963).

The Ozarks have been divided into two sections by Braun (1950): the Interior Highlands and the Forest-Prairie Transition. The Interior Highlands include Shannon and Carter counties and contain the bulk of the Ozark oak-hickory forest. In this area, oaks codominate with yellow or shortleaf pine (*Pinus echinata*) and locally may form pure stands (Braun, 1950). Mature stands of shortleaf pine occur in only a few localities in the southeastern Ozarks and the Missouri-Arkansas border (Critchfield and Little, 1966: map 42).

Steyermark (1940) further subdivides the Ozark oak-hickory forests into five edaphic associations based on physical, chemical, and local moisture conditions:

1. Sugar Maple-Bitternut Hickory Association (*Acer saccharum*-*Carya cordiformis*)
2. Sugar Maple-White Oak Association (*Acer saccharum*-*Quercus alba*)
3. Oak-Hickory Association (*Quercus*-*Carya*)
4. Oak-Pine Association (*Quercus*-*Pinus echinata*)
5. White Oak-Red Maple Association (*Quercus alba*-*Acer rubrum*)

All five associations occur within the study locality (Steyermark, 1940).

This description of the Ozark flora can only be applied as a generalization. In several areas of the Missouri Ozarks, prairie flora inhabit forest openings and glades (Braun, 1950; Steyermark, 1963). The distribution and composition of the Ozark's modern flora described by Steyermark

(1940, 1959, 1963) is probably not the same as it was at the advent of European settlement. Sauer (1950:59) reports the following changes in the Ozark forests:

1. greater density of stand and more undergrowth, resulting from the cutting of large timber and the cessation of fires;
2. a great decrease in the lowland forest area as a result of land clearance for farming; and
3. a relative increase of those species that have the most efficient means of propagation, such as oaks and elms with their coppicing habits and, in the bottoms, the sycamore and cottonwoods with wind-blown seeds.

Beilman and Brenner (1951a; 1951b) believe the Ozark oak-hickory forests are a relatively recent development and are still maturing. Steyermark (1959) disagrees, feeling that the present oak-hickory forests were derived from widespread mixed Tertiary forests as a result of decreased available moisture. The palynological studies by Mehringer and others (1968; 1970), King (1973), and King and Lindsay (1976) substantiate Beilman and Brenner's (1951a; 1951b) belief that the Ozark oak-hickory forests are a relatively recent development. King and Lindsay (1976) suggest that the present oak-hickory forests did not become established in the western Missouri Ozarks until after 10,000 B.P.

ARCHAEOLOGICAL OVERVIEW OF THE SOUTHEAST MISSOURI OZARKS

For archaeological purposes, Chapman (1975) divides Missouri into six general physiographic regions based on spatial, climatic, topographic, geologic, biologic, and aboriginal settlement patterns and cultural developments. The physiographic regions are: Southwest Drainage, Western Prairie, Ozark Highland, Northwest Prairie, Northeast Prairie, and Southeast Riverine (Fig. 12). These regions are then subdivided into stream drainages, which Chapman (1975) designates as localities (Fig. 13). The study area is located in the Current-Eleven Point Locality of the Southwest Drainage Region and the Upper Black-St. Francis locality of the Ozark Highland Region (Fig. 13).

The Southwest Drainage Region is composed of steep-sided hills and deeply incised narrow valleys, with closely spaced and meandering rivers. The soil is thin and rocky; 75% of the area has a greater than 5% slope. Vegetation consists of oak-shortleaf pine and oak-hickory forests with localities of prairie grasses and herbs (Chapman, 1975).

Medium-sloped hills with narrow-bottomed valleys characterize the Ozark Highland Region. The interfluvies are either broad and flat or sharp and narrow. Small prairies occur on the flat interfluvies and the upland forest vegetation is composed of oak-hickory, shortleaf pine, or mixed oak and pine stands. The soil is thin and infertile with a clay base (Chapman, 1975).

Fauna native to the study locality that were attractive to the early pioneers and the Indians who preceded them included deer (*Odocoileus virginianus*), bison (*Bison bison*), elk or wapiti (*Cervus elaphus*), wolves (*Canis lupus* and *C. niger*), bears (*Ursus* spp.), panther or mountain lion (*Felis concolor*), wild cats or bobcats (*Lynx rufus*), beaver (*Castor canadensis*), otter (*Lutra canadensis*), muskrat (*Ondatra zibethicus*), mink (*Mustela vison*), raccoon (*Procyon lotor*), opossum (*Didelphis virginiana*), skunk (*Mephitis mephitis* and *Spilogale putorius*), fox squirrel (*Sciurus niger*), gray squirrel (*Sciurus carolinensis*), fox (*Urocyon cinereoargenteus* and *Vulpes vulpes*), chipmunk (*Tamias striatus*), and cottontail rabbit (*Sylvilagus floridanus*). Turkey (*Me-*



Figure 12. Archaeological-physiographic regions of Missouri. Shannon and Carter counties are outlined. Adapted from Chapman, C.H., 1975.

Figure 13. Stream drainage localities of Missouri. Shannon and Carter counties are outlined. Adapted from Chapman, C.H., 1975.

Explanation

Northwest Prairie Region

- 1A. Tarkio, 1B. Nodaway, 1C. Platte, 1D. Grand,
1E. Chariton, 1F. Lower Missouri Valley I, 1G. Lamine.

Western Prairie Region

- 2A. Upper Osage.

Southwest Drainage Region

- 3A. Neosho, 3B. White, 3C. Current-Eleven Point.

Ozark Highland Region

- 4A. Lower Osage, 4B. Gasconade, 4C. Meramec,
4D. Upper Black-St. Francis, 4E. Castor-Whitewater.

Northeast Prairie Region

- 5A. Lower Missouri Valley II, 5B. Greater St. Louis,
5C. Cuivre, 5D. Salt, 5E. Wyaconda-Fabius,
5F. Des Moines, 5G. Mississippi Valley North.

Southeast Riverine Region

- 6A. Mississippi Valley Central, 6B. St. Francis Riverine,
6C. Bootheel Riverine.

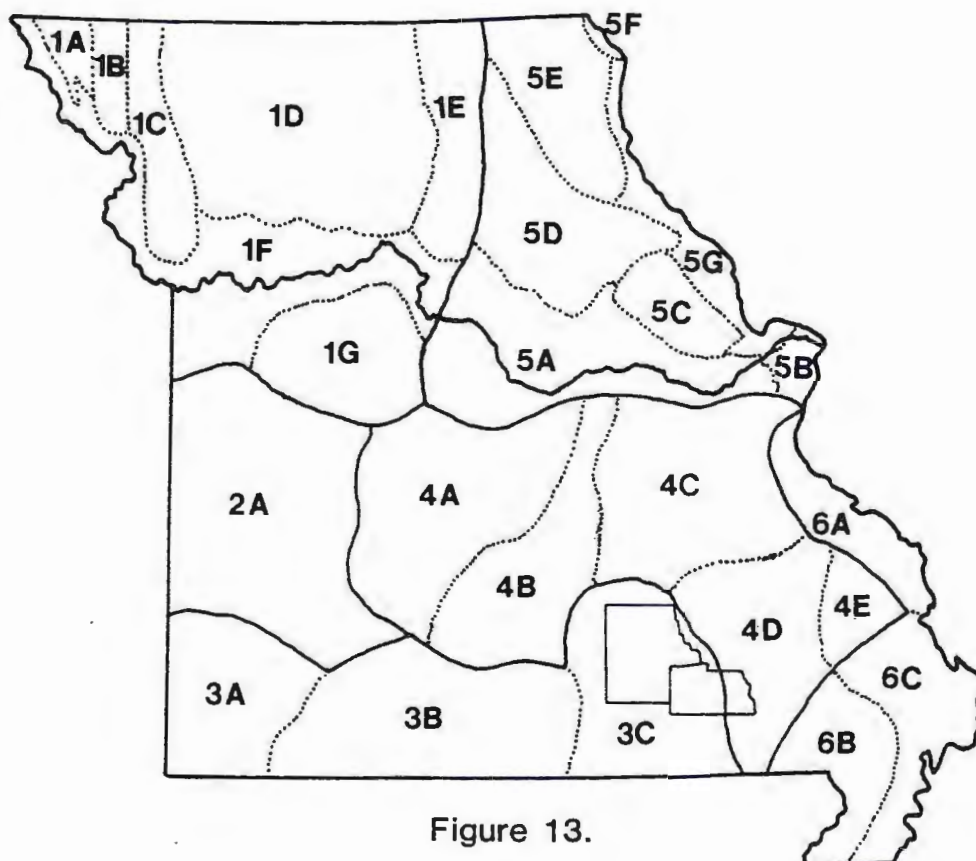


Figure 13.

leagris gallopavo), quail (*Colinus virginianus*), and passenger pigeon (*Ectopistes Migratorius*) were also important (Sauer, 1920; Chapman, 1975).

Fish in the area included bass (*Micropterus* spp.), jack salmon (wall-eyed pike) (*Stizostedion vitreum*), sunfish (*Lepomis* spp.), stone cat (*Noturus flavus*), suckers (Catostomidae), channel cat (*Ictalurus* spp.), bullhead cat (*Ictalurus* spp.), mud cat (*Ictalurus* spp.), buffalo (*Ictiobus* spp.), crappie (*Pomoxis* spp.), short-nosed gar (*Lepisosteus platostomus*), eel (*Anguilla rostrata*), and minnows (Cyprinidae) (Sauer, 1920; Chapman, 1975). Bullfrogs (*Rana catesbiana*) also occurred (Sauer, 1920).

Chapman (1975) combined tentative chronology and terminology for climatic episodes from Bryson, Baerreis, and Wendland (1970) and from Bryson and Wendland (1967) with archaeological periods to provide a chronological framework to discuss Missouri archaeology (Table 3). Radio-carbon dates have been converted to absolute time units B.C. by subtracting 1950, but are not calibrated (Chapman, 1975). The cultural traditions and archaeological periods used by Chapman (1975) are shown in Table 4.

UNSPECIALIZED HUNTER-GATHERER TRADITION

Early Man Period: ?-12,000 B.C. (?-13,950 B.P.)

There is no conclusive evidence that any human activity occurred in Missouri before 12,000 B.C. during the Early Man Period. However, material from the Kimmswick, Pomme de Terre, Grundel, and Miami mastodon sites suggests possible evidence of human presence during the Early Man Period (Chapman, 1975). None of these sites are in the study area.

EARLY HUNTER TRADITION

Paleo-Indian Period: 12,000-8000 B.C. (13,950-9950 B.P.)

In the Southwest Drainage and Ozark Highland regions, the only evidence of the Early Hunter Tradition of the Paleo-Indian Period is three fluted points found in Carter County. As a result of a colder climate during this period, Early Hunters probably had a limited supply of vegetable food

TABLE 3. CLIMATIC EPISODES AND PROPOSED CHRONOLOGICAL PERIODS
USED TO DISCUSS MISSOURI ARCHAEOLOGY

Climatic Episodes	Rough Date	Period	Absolute Date
Recent	A.D. 1970		1970
		Historic	
	A.D. 1850 ---		
Neo-Boreal			1700 -----
		Late Mississippi	
	A.D. 1550 ---		
Pacific II			
	A.D. 1450 ---		1450 -----
Pacific I		Middle Mississippi	
	A.D. 1200 ---		1200 -----
Neo-Atlantic		Early Mississippi	
	A.D. 900 ---		900 -----
Scandic		Late Woodland	
	A.D. 400 ---		400 -----
			A.D. 1 1 B.C.
		Middle Woodland	
Sub-Atlantic			500 -----
		Early Woodland	
	940 B.C. ---		
			1000 -----
Sub-Boreal		Late Archaic	
	2730 B.C. ---		
			3000 -----
Atlantic IV		Middle Archaic	
	4030 B.C. ---		
			5000 -----
Atlantic III			
	5100 B.C. ---		
Atlantic II		Early Archaic	
	5780 B.C. ---		
Atlantic I			
	6500 B.C. ---		
			7000 -----
Boreal II		Dalton	
	7190 B.C. ---		
Boreal I			
	7700 B.C. ---		
			8000 -----
		Paleoindian	
Pre-Boreal			12,000 -----
		Early Man	

From Chapman, C.H., 1975.

TABLE 4. CULTURAL TRADITIONS IN RELATION TO ARCHAEOLOGICAL PERIODS

TRADITION	PERIOD
Village Farmer	Late Mississippi A.D. 1450-1700 Middle Mississippi A.D. 1200-1450 Early Mississippi A.D. 900-1200
Prairie-Forest Potter	Late Woodland A.D. 400-900 Middle Woodland 500 B.C.-A.D. 400 Early Woodland 1000-500 B.C.
Forager	Late Archaic 3000-1000 B.C. Middle Archaic 5000-3000 B.C. Early Archaic 7000-5000 B.C.
Hunter-Forager	Dalton 8000-7000 B.C.
Early Hunter	Paleoindian 12,000-8000 B.C.
Unspecialized Hunter-Gatherer	Early Man (?) -12,000 B.C.

From Chapman, C.H., 1980

available and concentrated primarily on obtaining large-to-moderately large game animals. Small game was also included as part of their subsistence (Chapman, 1975).

HUNTER-FORAGER TRADITION

Dalton Period: 8000-7000 B.C. (9950-8950 B.P.)

Chapman (1975) suggests that Early Hunters evolved to the Hunter Forager Tradition as the climate changed from glacial to postglacial. It is thought that the Dalton Period was one of change from a nomadic hunting subsistence to a seminomadic foraging subsistence (Chapman, 1975).

In the Southwest Drainage Region, Dalton Serrated points in conjunction with foraging implements have been found in the lowest levels of the Standlee and Rice shelters. Several Dalton Serrated points have been found on the surface of sites along the Current River in Shannon County (Chapman, 1975). In the Ozark Highland Region, material from Rodgers Shelter suggests that Dalton Period occupants obtained a variety of large and small game and that hickory nuts and black walnuts were part of the subsistence base (McMillan, 1976b). Sandstone mortars, manos, and hammerstones associated with some Dalton sites indicate that nut, berry, and seed collecting were part of the subsistence pattern (Chapman, 1975).

FORAGER TRADITION

Early Archaic Period: 7000-5000 B.C. (8950-6950 B.P.)

During the Early Archaic Period for the Forager Tradition, subsistence activities began to change and a greater number of ecological niches were exploited. Hunting and gathering continued to be the major economic activity, although more emphasis was placed on fishing and collecting shellfish. At this time, vegetable foods became important in the subsistence economy and camps were used as a base for collecting activities (Chapman, 1975).

No evidence of the Early Archaic Period has been found in the Current-Eleven Point Locality of the study area. The only indications of Early Archaic occurrence in the Upper Black-St. Francis Locality are a Dalton Serrated and a Rice Lanceolate associated with a variety of other chipped stone artifacts (Chapman, 1975).

Middle Archaic Period: 5000-3000 B.C. (6950-4950 B.P.)

The Middle Archaic Period began when the climate became warmer and dryer throughout much of the central United States and lasted until the dry period reached its height. Hunting and trapping of small animals together with the use of plants, especially nuts and seeds, continued to be of great importance during this period. Archaeological evidence for the first use of twined-fiber fabrics for a variety of items occurs at this time (Chapman, 1975).

At Rodgers Shelter two different subsistence patterns, designated Middle Archaic Period I and II, were found for the Middle Archaic Period. During Middle Archaic Period I, deer remains decrease sharply. Although bison bones were not found in large quantities, their presence indicates their continued importance. Prairie fauna seemed to be obtained only incidentally, with a greater emphasis on the procurement of small game. Squirrels were especially important. Hickory nuts, walnuts, and blackberries were found at what was probably the same zone (McMillan, 1976b).

In Middle Archaic Period II, emphasis shifted to rabbits and other small rodents and away from squirrels. Freshwater mussels also became important. Plant processing, such as complex crushing and grinding, became less important (McMillan, 1976b).

Chapman (1975) does not mention the presence of any Middle Archaic Period sites in either the Current-Eleven Point or Upper Black-St. Francis localities.

Late Archaic Period: 3000-1000 B.C. (4950-2950 B.P.)

The beginning of the Late Archaic Period was probably very dry and was a time of adaptation to ecological niches not utilized by earlier Foragers. Prairies had expanded eastward and forests had diminished, along with the plants and animals that served as food supplies (Chapman, 1975).

According to Chapman (1975) the prairie potato (*Psoralea* spp.) may have been an important food at this time as well as other pulpy plants such as the cambium layer of elm. This is indicated by the many digging and grinding tools found associated with late Archaic sites in the Western Prairie, the northern Ozark Highland, and Northeast Prairie regions (Chapman, 1975).

At Rodgers Shelter great emphasis was placed on deer hunting during the Late Archaic. Exploitation of turtles and mussels also increased at this time (McMillan, 1976b).

No sites identified as Forager Tradition have been found in the Current-Eleven Point Locality, nor in the Upper Black-St. Francis Locality. In the Ozark Highland, Foragers of the Middle Archaic continued to live through the Late Archaic Period without much change (Chapman, 1975).

PRAIRIE-FOREST POTTER TRADITION

Early Woodland Period: 1000-500 B.C. (2950-2450 B.P.)

Gardening is often assumed to distinguish the Late Archaic Period from the Early Woodland Period. Squash (*Cucurbita* spp.), gourds (*Cucurbita* spp.), and possibly marsh elder (*Iva* spp.), sunflowers (*Helianthus* spp.), goosefoot (*Chenopodium* spp.), and canary grass (*Phalaris* spp.) were cultivated during the Late Archaic but there is insufficient evidence to indicate horticulture as the basis for cultural change from the Archaic to the Woodland Period (Chapman, 1980).

There are indications that the Forager Tradition continued into the Early Woodland Period in the Southwest Drainage and Ozark Highland regions (Chapman, 1980). Chapman (1980) suggests that Indians of the Early Woodland Period in the Southwest Drainage Region had a subsistence pattern similar to that of the Osage Indians. They hunted bear (*Ursus* spp.), elk (*Cervus elaphus*), deer (*Odocoileus virginianus*), buffalo (*Bison bison*), and trapped beaver (*Castor canadensis*). Chapman (1980) also suggests that they gathered persimmons (*Diospyrous virginiana*), nuts, and berries. At Boney Spring in the Ozark Highland Region, an Early Woodland storage pit contained a nutting stone as well as walnuts (*Juglans nigra*), hazelnuts (*Corylus americana*), hickory nuts (*Carya* spp.), and acorns (*Quercus alba* and *Q. macrocarpa*) (King and McMillan, 1975). They

also found bulrush (*Scirpus* spp.), sedge (*Carex* spp.), knotweed (*Polygonum* spp.), pokeberry (*Phytolacca americana*), buttercup (*Ranunculus* spp.), blackberry (*Rubus* spp.), wild plum (*Prunus* cf. *americana*), wild grape (*Vitis* spp.), violet (*Viola* spp.), dogwood (*Cornus* spp.), elderberry (*Sambucus canadensis*), giant ragweed (*Ambrosia trifida*), cocklebur (*Xanthium* spp.), and squash (*Cucurbita pepo*) seeds.

Middle Woodland Period: 500 B.C.-A.D. 400 (2450-1550 B.P.)

During the Middle Woodland Period pottery manufacture, corn horticulture, burial-mound construction, and widespread trading became an important part of the Prairie-Forest Potter Tradition (Chapman, 1980). The Forager Tradition continued into the Middle Woodland Period in the Current-Eleven Point Locality of the Southwest Drainage Region.

The Round Spring Site (part of this study) on the Current River has a Woodland component that may have existed as early as 500 B.C. (Lynott, 1981a). No Middle Woodland data have been found in the Upper Black-St. Francis Locality, although during this period evidence from the surrounding area indicates that chenopods (*Chenopodium* spp.) and hickory nuts were harvested along the bottomlands.

Late Woodland Period: A.D. 400-900 (1550-1050 B.P.)

The bow and arrow had become the major weapon for hunting and warfare by the beginning of this period. Although the earlier Prairie-Forest Potter Tradition economy continued, there was a movement away from gardening and specialized resource exploitation to a more generalized hunting and gathering subsistence (Chapman, 1980).

In the Current-Eleven Point Locality, Prairie-Forest Potter Tradition intrusions occurred (Chapman, 1980). At Rodgers Shelter in the Ozark Highlands, hunting of deer and turkey was again important as was fishing and mussel collecting (McMillan, 1976b).

VILLAGE FARMER TRADITION

Early Mississippi Period: A.D. 900-1200 (1050-750 B.P.)

Village farming along rivers and efficient exploitation of natural resources characterize the Early Mississippi Period. Fish, shellfish, waterfowl, amphibians, and water-dwelling mammals were obtained from the rivers. Fruits and seeds of trees, shrubs, and herbs were also exploited as part of the subsistence base. The villages were within or near fortified civic-ceremonial centers that contained platform mounds arranged around a courtyard or plaza. Ceremony and mound building became important aspects of this period (Chapman, 1980).

The Loftin Site in the Southwest Drainage Region contained an assemblage of farming implements such as digging tools, manos, metates, and adzes. Corn and squash have been found at several sites in the Ozark Highland Region (Chapman, 1980).

In the Current-Eleven Point Locality, two Early Mississippi sites are known. The Pigmen Mound site is located in the Eleven Point Drainage. It is the only known Mississippian temple mound in the eastern Ozarks. Two artifact assemblages were found at this site, a Transitional Woodland/Mississippi component and an Early Mississippi component representing the Naylor Phase (Lynott, 1980).

The Gooseneck Site (part of this study) on the Current River is also an Early Mississippi Naylor Phase site; it is small and may represent an early Mississippian hamlet. White-tailed deer (*Odocoileus virginianus*), opossum (*Didelphis virginiana*), eastern mole (*Scalopus aquaticus*), eastern cottontail (*Sylvilagus floridanus*), gray squirrel (*Sciurus niger*), fox squirrel (*Sciurus niger*), muskrat (*Ondatra zibethicus*), beaver (*Castor canadensis*), gray fox (*Urocyon cinereoargenteus*), and racoon (*Procyon lotor*) bones have been found there. Fish found at the site include bowfin (*Amia calva*), hog sucker (*Hypentelium nigricans*), sucker (*Catostomidae*), and rock bass (*Ambloplites rupestris*). Turtle (Chelonia), ducks (Anatidae), and turkey (*Meleagris gallapavo*) were also found. Carbonized hickory (*Carya* spp.) nut shells, acorn (*Quercus* spp.) shells, and grape (*Vi-*

tis spp.), sumac (*Rhus* spp.), and knotweed (*Polygonum abiculare*) seeds have been found at the Gooseneck Site. One cupule of corn (*Zea mays*) was also found (Potter, 1973; Lynott, 1980).

The Round Spring Site dates to about A.D. 1200, which places it at the end of the Early Mississippi Period or at the beginning of the Middle Mississippi Period. The site has a cemetery and probable village component (Lynott, 1981b).

Middle Mississippi Period: A.D. 1200-1450 (750-500 B.P.)

During the Middle Mississippi Period, village farming and resource exploitation continued in a fashion similar to that of the Early Mississippi Period, but it was a time of population growth and territorial expansion under control of the civic-ceremonial center. Mounds grew larger in size and arts and crafts achieved new heights as the Village Farmer Tradition reached its classic development (Chapman, 1980).

The Gypsy Joint site in the Upper Black-St. Francis Locality belongs to the Middle Mississippi Period and is part of the Powers Phase. More than 1600 grams of plant material were recovered here. The bulk of the plant material (1576 g) was nutshells. Forty grams of corn kernels and cob fragments were recovered and approximately 20,000 seeds. Hickory nuts are the most abundant and widespread plant remains found at the Gypsy Joint site. Acorns and walnuts were present in small quantities (Smith, 1978).

Corn (*Zea mays*), marsh elder (*Iva* spp.), and possibly sunflower (*Helianthus* spp.) represent the cultigens found at the site. Corn was probably the major food source, although hickory nuts may also have played a significant part in subsistence. Knotweed and chenopod seed make up the majority of the 20,000 seeds found, suggesting that they were harvested for food. Wild bean (*Phaseolus* spp.), grape (*Vitis* spp.), crab apple (*Pyrus* spp.), and plum (*Prunus* spp.) or cherry (*Prunus* spp.) were found in small quantities and were probably minor supplements to subsistence. The six morning glory (*Ipomoea* spp.) seeds found were probably growing as garden weeds and were preserved at the site by chance (Chapman, 1980).

The Southwest Drainage Region appears to have been abandoned during the Middle Mississippi Period although the area was used for hunting and foraging (Chapman, 1980).

Late Mississippi Period: A.D. 1450-1700 (500-250 B.P.)

The Late Mississippi Period has also been called the Protohistoric Period (Chapman and Chapman, 1964). During this time, dispersal and abandonment of most cultural centers occurred north of the junction of the Ohio and Mississippi rivers (Chapman, 1980).

The advent of European influence on the area (direct or indirect contact by DeSoto in A.D. 1540) probably brought about part of the decline of the Village Farmer Tradition. European diseases were introduced and in some areas DeSoto disposed of town leaders (Chapman and Chapman, 1964). For whatever reasons, large towns were abandoned or reduced and decentralization occurred.

The Oneota were the Protohistoric Indians of Missouri. They hunted deer, elk, bison, turkey, and waterfowl. Fishing was also important as was turtle catching and mussel collecting. Corn, beans, and squash were cultivated. The Oneota merge into the historic Indian cultures of Missouri without a distinct boundary (Chapman and Chapman, 1964).

Historic Period: A.D. 1700-Present (250 B.P.-Present)

The Osage tribe was one of the major historic groups of native Indians, occupying the Ozark Highland and western prairie. Most of the central part of Osage territory has been inundated by the waters backed up by Bagnal Dam, so it has not been archaeologically investigated (Chapman, 1952). Cultural objects of the Osage show a relationship to the Oneota (Chapman, 1952; Chapman and Chapman, 1964).

Hunting was the main means of subsistence. Bear, bison, deer, and beaver were important game. Cultivation of corn, beans, and squash was also important (Chapman, 1952; Chapman and Chapman, 1964). These foods were supplemented by the gathering of persimmons, nuts, and lotus (*Nelumbo lutea*) roots. Lotus seeds were also eaten (Chapman and Chapman, 1964).

The Missouri tribe appears to be affiliated with the Oneota culture. Hunting was paramount but they were horticulturalists also. The villages were large and served as permanent bases from which to hunt (Chapman, 1952; Chapman and Chapman, 1964). Bison, deer, elk, bear, and beaver were important game animals (Chapman, 1952). Turkey and duck were also procured as well as fish and turtle (Chapman and Chapman, 1964). Corn, beans, and squash were cultivated (Chapman, 1952; Chapman and Chapman, 1964). Seeds, nuts, roots, and berries supplemented their subsistence (Chapman, 1952).

Table 5 summarizes the chronological sequences of the cultural developments in the study area. The sequence is based on archaeological evidence found in the Current-Eleven Point and Upper Black-St. Francis localities.

TABLE 5. CHRONOLOGICAL SEQUENCE OF CULTURAL
DEVELOPMENTS IN THE STUDY AREA

		Southwest Drainage Region	Ozark Highland Region
Period	Date	Current- Eleven Point Locality	Upper Black- St. Francis Locality
Middle Mississippi	A.D. 1450	Pigmen Mound?	Powers phase?
	A.D. 1200		
Early Mississippi		Pigmen Mound	Transitional Prairie- Forest Potter and Village Farmer
	A.D. 900		
Late Woodland		Prairie-Forest Potter Tradition	Wappapello Lake aggregate
	A.D. 400		
Middle Woodland		Forager Tradition continues	No data
	500 B.C.		
Early Woodland		Forager Tradition	Forager Tradition
	1000 B.C.		
Late Archaic		No data	No data
	3000 B.C.		
Middle Archaic		No data	No data
	5000 B.C.		
Early Archaic		Rice Complex (?)	Questionable evidence Dalton Serrated and Rice Lanceolate
	7000 B.C.		
Dalton		Scattered Dalton Serrated	Cache of Dalton Serrated
	8000 B.C.		
Paleoindian		Scattered Clovis Fluted	No data
	12,000 B.C.		
Early Man		No data	No data
	?		

Adapted from Chapman, C.H., 1978, 1980.

SPECIFIC SITE DESCRIPTIONS

BUTTONBUSH BOG

Buttonbush Bog (Fig. 1) is located in the NW 1/4, SW 1/4, NE 1/4 of section 32, T27N, R3W, of the Birch Tree 15' quadrangle, in Shannon County, at approximately 90°18' W long., and 36°58' N lat. Located 5 km southeast of Winona, the site is in the Winona Ranger District of the Mark Twain National Forest, at an elevation of 297 m above sea level. The bog is contained within a small sinkhole (100 m in diameter) on a dolomitic ridge of the Roubidoux formation. Open water occurs only in a small area in the center of the bog, although a local resident (oral commun., 1980) reported that until 1975 the entire bog surface was covered by shallow water. *Cephalanthus occidentalis* var. *occidentalis* f. *occidentalis* (buttonbush) is the dominant vegetation growing on the bog surface (Fig. 14). *Polygonum* sp., *Nuphar luteum*, *Polygonum hydropiperoides*, and *Poa* sp. were also found growing in the bog (Appendix II.A.). Although no diagnostic archaeological material has been found at Buttonbush Bog, several chert waste flakes have been found around the perimeter.

ROUND SPRING SHELTER, ROUND SPRING SITE 23SH19

Round Spring Shelter (approximately 91°24'30" W long., 37°17' N lat.) is in the NW 1/4, SW 1/4, NW 1/4 of section 20, T30N, R4W, of the Round Spring 7.5' quadrangle, Shannon County (Fig. 1), with an elevation of 207 m. The shelter (Fig. 15) is a small cave located in the southeast side of the Round Spring sinkhole. Round Spring is located 16 km northwest of Eminence in the Ozark National Scenic Riverways. The sinkhole is a nearly circular basin 20 m in diameter, formed when a cavern roof in the Eminence dolomite collapsed. Water from the spring (Fig. 16) flows under a natural bridge formed by the remaining part of the cavern roof, into the Current River (Vineyard and Feder, 1974; revised ed., 1982).

Round Spring Shelter is part of Round Spring Site 23SH19 (Fig. 17) where archaeological investigations were carried out in 1981 by the Midwest Archaeological Center, Lincoln, Nebraska. As part of the 1981 investigation, a 50 x 50 cm test unit was excavated near the front of the shelter



Figure 14. Photograph of Buttonbush Bog. Photo by C.R. Huber, 1981.

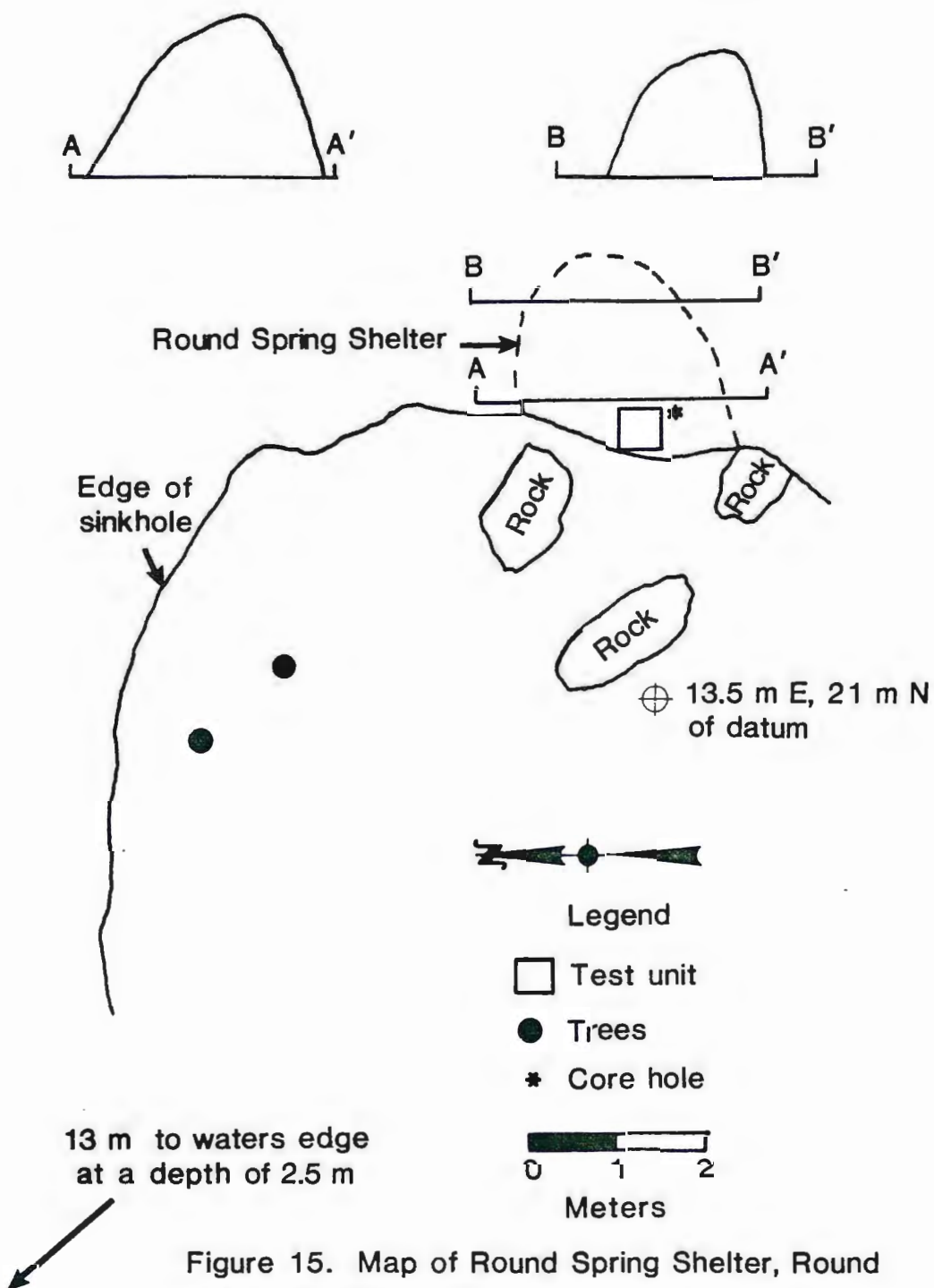


Figure 15. Map of Round Spring Shelter, Round Spring Site 23CT54. Redrawn from National Park Service, 1981.



Figure 16. Photograph of Round Spring. Photo by author, 1980.

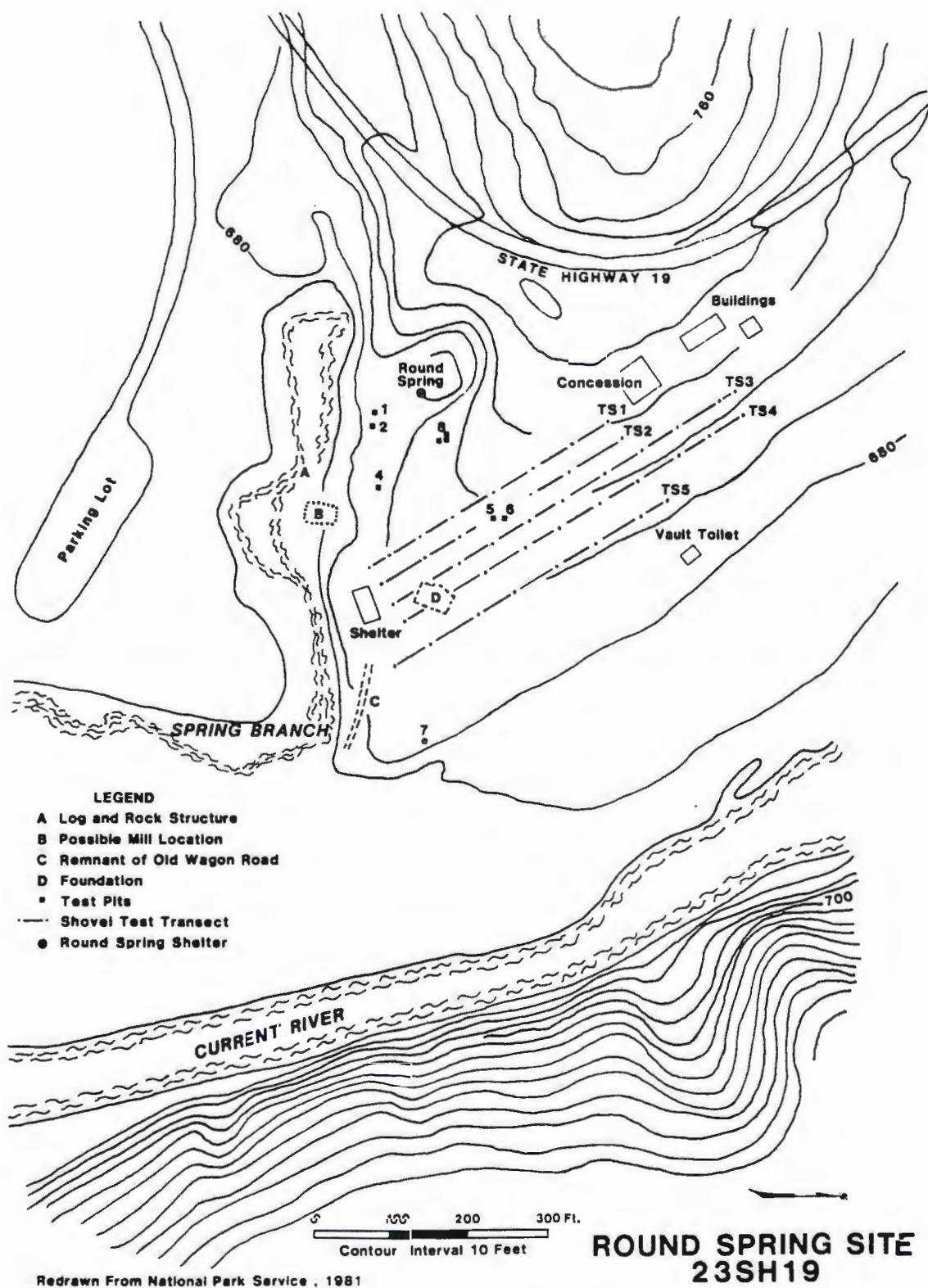


Figure 17. Map of Round Spring Site 23SH19.

(Fig. 15) to a depth of 80 cm. Ceramics, lithic tools and debris, bone, and shell were recovered from the test square (Lynott, written commun., 1981).

GOOSENECK SITE 23CT54

Gooseneck Site 23CT54 (Fig. 1) is in the E 1/2, SW 1/4, and SW 1/4 of section 15, T25N, R1E, of the Grandin SW 7.5' quadrangle in Carter County at an elevation of 128 m. The archaeological site is at approximately 90°57' W long., 36°49' N lat., 12 km southeast of Eastwood in Hawes Memorial Campground, Ozark National Scenic Riverways, on the Carter/Ripley County line. The Gooseneck site (Fig. 18) is located on the second terrace on the west bank of the Current River and is underlain by the Gasconade formation. The site was investigated in 1971 by the University of Michigan field school, and again in 1979 by the National Park Service.

OZARK SINK POND

One of the sites investigated was an unnamed sinkhole pond that will be referred to as Ozark Sink Pond (Fig. 1) for convenience. The pond (at approximately 90°03' W long. and 36°55'30'' N lat.) is in the SE 1/4, SE 1/4, S/W 1/4 of section 10, T26N, R1W, of the Van Buren South 7.5' quadrangle in Carter County and is not shown on the topographic map. Located 8 km southeast of Van Buren, the site is within the Van Buren Ranger District of the Mark Twain National Forest, at an elevation of 268 m above sea level. The pond is small (approximately 30 x 50 m), occurs on a ridge of dolomitic Roubidoux formation, and has a maximum water depth of 35 cm. *Cephalanthus* (buttonbush) is abundant around the edge of the pond with *Eleocharis obtusa* var. *obtusa*, and *Carex* spp. growing in the pond (Appendix II.B). Several waste flakes, but no diagnostic archaeological material, have been found at the site.

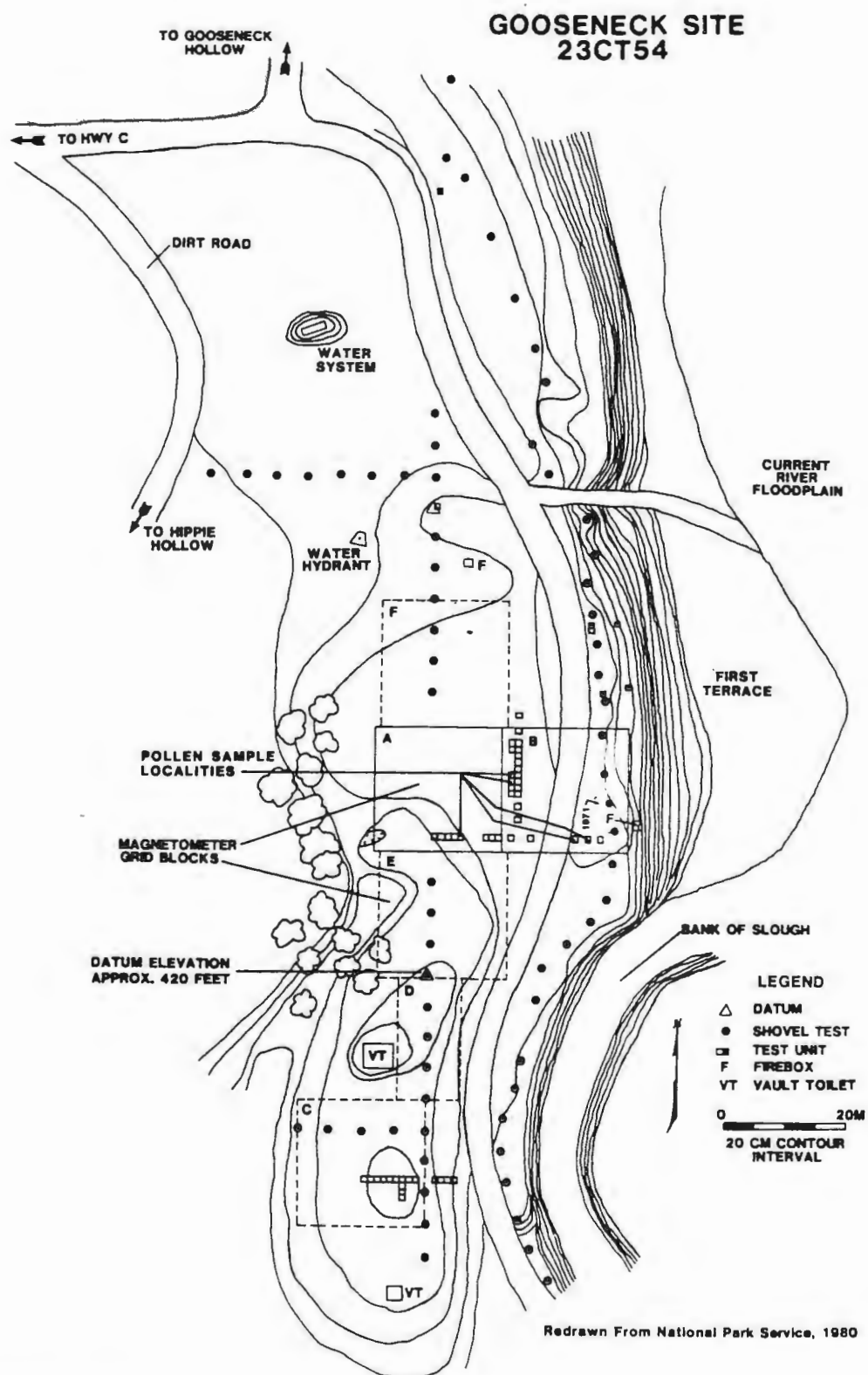


Figure 18. Map of Gooseneck Site 23CT54.

INCIDENTAL SITES

Three incidental sites were visited either to collect plant specimens known to grow in the area but not at the specific sites investigated, or to collect plants from restricted habitats. The sites are: a cypress swamp, 3 km east of Naylor at the Powers Fort Archaeological Site; Lewis Lake, 6 km north of Winona on the west side of Highway 19; and Highway 60, 5 km west of Van Buren.

METHODS

CORING METHODS AND COLLECTION OF ARCHAEOLOGICAL SEDIMENTS

Buttonbush Bog

Buttonbush Bog was cored in August 1981 with a Livingstone piston sampler 5 cm in diameter and 100 cm long (Livingstone, 1955). The 302-cm core was extracted from the approximate center of the bog. The core was extruded, measured, described, and then wrapped with Saran Wrap and aluminum foil. Upon return to the laboratory, the core was stored in a constant temperature cooler at 4°C (Moore and Webb, 1978) to prevent mold and fungus growth.

Round Spring Shelter, Round Spring Site, 23SH19

Round Spring Shelter was cored in July 1981 with a glacial bucket corer, 10 cm from the SE corner of the test square previously mentioned. As a result of the shelter's limited confines, the difference in sediment consolidation, and obstructions encountered while coring; it was not possible to extract equal sample intervals. At 26 cm, a rock was encountered, necessitating enlargement of the core hole on one side to core past the obstruction. This resulted in a 4-cm overlap in sampling between 19 and 23 cm. The bedrock floor of the shelter or rock debris was encountered at 85 cm, 5 cm below the depth archaeologists reached when excavating a small test pit in the shelter (Lynott, 1981, written commun.). The sediment samples from Round Spring Shelter were removed from the corer and put in plastic sample bags. Each bag was sealed in the field and upon return to the laboratory was refrigerated at 4°C to prevent the growth of mold and fungus.

Gooseneck Site 23CT54

Eight archaeological soil samples collected during the 1979 field season were obtained from Mark J. Lynott, National Park Service, Midwest Archaeological Center, Lincoln, Nebraska. The soil samples were collected with trowels from the bottom of arbitrary (10 cm) excavation levels, and were sealed in plastic sample bags. Four of the samples are from the midden area along the terrace edge, two are from a feature that is believed to be a house, and two are from an A-P and subsoil

horizon (Lynott, 1981, written commun.). The excavation test squares from which the samples were collected are shown in Figure 18.

Ozark Sink Pond

In July 1981, Ozark Sink Pond was cored at the approximate center with a Dutch auger. Five-centimeter samples were extracted to a depth of 245 cm, where gravel was encountered, making further penetration impossible. The samples were bagged in whirl pack bags and refrigerated at 4°C until they could be dried at room temperature (24°C). The sediment is composed of dolomitic silt and clay with little organic matter. Preliminary analysis of the sediment indicated little pollen content and the samples were not analyzed.

COLLECTION OF MODERN POLLEN RAIN SAMPLES

As part of this study, several bryophytic polsters and pinch samples were analyzed for modern Ozark pollen rain. Two bryophytic polsters and three pinch samples representing pine-oak forests were collected in August 1981. The bryophytic polsters were collected near Ozark Sink Pond in Carter County. The three pinch samples were collected, one each at Buttonbush Bog and Round Spring Site 23SH19 in Shannon County and one at Gooseneck Site 23CT54 in Carter County. In addition, five bryophytic polsters were collected in August 1982 in the vicinity of Buttonbush Bog.

At the time of collection, the bryophytic polsters were sealed in sterile collecting bags. Upon return to the laboratory, the 1981 samples were dried to prevent the growth of mold and fungal development. The 1982 samples were processed immediately after returning to the laboratory.

Pinch samples were collected by walking the field area near the sites and scooping up small surface samples with a spoon. A plastic sample bag of material weighing approximately one kilogram was collected at each site. This is a modification of the technique described by Lewis and Ogden (1965), who suggest collecting the upper centimeter of the soil surface in small vials. Each bag was sealed in the field and upon return to the laboratory was refrigerated at 4°C to prevent the growth of mold and fungus.

POLLEN ANALYTICAL METHODS

Buttonbush Bog

The core was systematically sampled at 4-cm intervals and processed for pollen, using a modified Faegri and Iverson (1975) technique (addition of KOH, HCl, HF, and acetolysis). Two *Eucalyptus* tablets containing $13,550 \pm 220$ grains were added to each sample so that pollen concentrations could be determined following a modification of Maher's (1972) technique. In addition, the pollen samples were washed through 10-micrometer mesh Nitex screens to remove clay, as described by Cwynar, Burden, and McAndrews (1979). (See Appendix III.A for the complete pollen processing procedure.) The samples were then stored in silicone oil (1000 centistokes at 29°C) for later counting.

Round Spring Shelter, Round Spring Site, 23SH19

The sediment samples from Round Spring Shelter were dried at room temperature and split through a Carpcor riffle box splitter according to Shackley (1975), until a representative sample of 30 g was attained. The samples were processed using a modified Faegri and Iverson (1975) technique (KOH, HCl, HF, and acetolysis). Only one *Eucalyptus* tablet was added to each sample (because of the low concentration of pollen in the sediments) for pollen concentration determination using a modification of Maher's (1972) technique. To reduce the initial volume of the samples, they were first washed through 20-mesh (0.833 mm) and 80-mesh (0.177 mm) screens. The samples were then concentrated and treated with HCl to remove carbonates. Next, the samples were sieved through 10-micrometer Nitex screens to remove clays as described by Cwynar and others (1979). A heavy liquid mixture of tetrabromethane and absolute ethanol (specific gravity 2.0) was then used to separate the pollen from the silt-sized material. This method is similar to the bromoform flotation technique of Moore and Webb (1978). The pollen samples were then processed using KOH, HF, and acetolysis, and stored in silicone oil (1000 centistokes at 29°C) for later counting. (See Appendix III.B for the complete processing procedure.)

Gooseneck Site 23CT54

Pollen extraction for the sediment samples was undertaken in the same manner described for the samples from Round Spring Shelter. (For a detailed description of the processing procedure, see Appendix III.B)

Modern Pollen Rain Samples

In preparation for pollen extraction, the bryophytic polsters were placed in two-liter plastic bottles. The bottles were then filled with an approximately 50/50 mixture of ethanol and distilled water. This is a modification of the techniques of Carrol (1943) and of Kapp (1969) who suggested the use of alcohol, and of Hansen (1949), who used water. The polsters were then agitated (Carroll, 1943; Hansen, 1949) for 15 minutes on a roller agitator to dislodge the pollen. The supernatant and polsters were then washed with distilled water through a Coors buchner filtering funnel (effective diameter of holes: 1000 micrometers) and a Coors filtering crucible (effective diameter of holes: 500 micrometers) to remove the large organic material. The liquid was collected in a large beaker and then centrifuged to concentrate the pollen residue. All of the polsters collected contained some substrate. Therefore, a modified Faegri and Iverson (1975) technique (addition of KOH, HCl, HF, and acetolysis) was used to get rid of carbonate and silicate substrate. In addition, the pollen samples were washed through 10-micrometer mesh Nitex screens to remove clay as described by Cwynar and others (1979). (See Appendix III.C for the complete procedure.) The samples were then stored in silicone oil (1000 centistokes at 29°C) for later counting.

Pollen extraction for the three pinch samples followed the same technique used for the Round Spring Shelter sediment samples and is described in detail in Appendix III.B. Pollen concentration was not attempted on the modern pollen rain samples; therefore no *Eucalyptus* tablets were added to the samples.

PALYNOMORPH IDENTIFICATION AND COUNTING TECHNIQUES

Processed samples were mounted on glass slides, using 22 x 30 mm cover slips. A minimum of 400 grains were identified and counted as the quantity referred to as the "pollen sum." The pollen sum includes trees, shrubs, and herbs (including vascular cryptogams). Indeterminable and unknown pollen grains, aquatic pollen, algae, fungal spores, and *Eucalyptus* spike grains were counted but not included in the 400 grains. Fungal spores were not counted in the modern pollen rain samples.

Pollen and spores were identified using the keys in Erdtman (1952, 1954), Maloney (1961), Kapp (1969), McAndrews, Berti, and Norris (1973), Faegri and Iversen (1975), and Moore and Webb (1978), and by comparison to the pollen reference collection at the Archaeometry Laboratory, University of Minnesota, Duluth. Identifications were made to the lowest taxonomic level possible. The degree of certainty of identification for some taxa is indicated by the use of "type" and "cf." The use of "type" indicates that the pollen grain matches not only that taxon but others also. An identification preceded by "cf." was uncertain as a result of inadequate reference material, poor preservation, or ill-defined morphology (Watts and Winter, 1966). Most of the spores identified are prefaced by "cf." usually as a result of inadequate reference material. Indeterminable grains were divided into five categories as proposed by Cushing (1967): broken, concealed, corroded, crumpled, and degraded. Well-preserved pollen grains that were not identified are expressed simply as unknowns.

Pollen counting was done on a Leitz Ortholux microscope equipped with a Bausch and Lomb 45x objective and a Leitz 100x oil-immersion objective. The microscope is equipped with Leitz periplan 10x oculars and an internal lens for total magnification of 560x and 1625x, respectively. Routine pollen identifications were made at 560x. Critical identifications were made using oil immersion in Anisole. The first transect was counted two millimeters from the south edge of the slide. Subsequent spacing of the transects was estimated to cover the slide at approximately equal intervals from south to north. The interval estimations were done to minimize errors associated

with nonrandom distribution of pollen grains. When the 400 sum was reached, pollen counts were continued to the end of the transect, thus completing the count. The slide was then sealed and placed on permanent file at the Archaeometry Laboratory, University of Minnesota, Duluth. The original copies of the pollen count sheets are also on file at the Archaeometry Laboratory, UMD.

STATISTICAL ANALYSIS AND DATA PROCESSING

Pollen count data were processed by a Control Data Corporation Cyber 815 computer at the University of Minnesota, Duluth, using the FORTRAN V program POLPERC (Caballero and Huber, 1983a; revised 1985). This program divides the total fossil palynomorphs into the categories of trees, shrubs, herbs, vascular cryptogams, unknowns, indeterminables, aquatics, mosses, algae, fungal spores, and pre-Quaternary microfossils. It then calculates these groups and the taxa within each group in terms of percentages of the divisions shown in Table 6. Original copies of the POLPERC tabulated data are on file at the Archaeometry Laboratory, UMD.

TABLE 6: POLPERC POLLEN PROGRAM DIVISIONS FOR PERCENTAGES

Trees + Shrubs + Herbs (Herbs + Vascular Cryptogams) = Pollen Sum
Unknowns and Indeterminables = Pollen Sum + Unknowns and Indeterminables
Aquatics = Pollen Sum + Aquatics
Mosses = Pollen Sum + Mosses
Algae = Pollen Sum + Algae
Fungal Spores = Pollen Sum + Fungal Spores
Pre-Quaternary Microfossils = Pollen Sum + Pre-Quaternary Microfossils

POLPERC also calculates confidence intervals related to these divisions using the following formula according to Mosiman (1965), Maher (1972), and Moore and Webb (1978):

95% confidence limit

$$= \frac{\hat{P} + [(1.96)^2/(2n)] \pm (1.96)(\sqrt{[\hat{P}(1 - \hat{P})n] + [(1.96)^2/(4n^2)]}}{1 + [(1.96)^2/n]}$$

Where:

$$\hat{P} = \frac{x}{n}$$

\hat{P} = Estimation of the true proportion of the pollen type within the pollen sum

x = Number of grains of the pollen type

n = Pollen sum (varies according to division: see Table 6)

When expressed as a percentage diagram, the confidence intervals can be used as a method of testing quickly for significant differences between samples (Maher, 1972). Maher (1972:536) states that:

"In general, if the point estimate of the proportion or ratio of either sample is included in the 0.95 confidence interval of the other, the two samples will not be found to differ significantly at the 0.05 level. But if the point estimate of neither of the samples is included in the 0.95 confidence interval of the other, the two samples usually will be found to differ significantly at the 0.05 level."

Eucalyptus pollen tablets containing $13,550 \pm 220$ grains were added to the samples so that pollen concentrations could be determined following a modification of Maher's (1972) technique.

Pollen concentrations were computed using this formula:

$$C = \frac{TE}{CE} \times \frac{CG}{V}$$

Where:

C = Concentration

TE = The number of *Eucalyptus* grains in the tablet

CE = The number of *Eucalyptus* grains counted on a slide

V = The volume of sediment processed in milliliters

CG = The number of grains of a specific taxa counted on a slide

DIAGRAM CONSTRUCTION

The pollen diagrams were drawn on a Zeta 3653x plotter made by Nicolet Zeta Corporation, Concord, California using the FORTRAN V program DRAWDIA (Caballero and Huber, 1983b; revised 1985). Sediment accumulation rate, loss-on-ignition, pollen sum and total fossil palynomorph (concentration and influx), and clastic sediment diagrams were also drawn using this program. A total of 20 different diagrams were drawn using the Zeta plotter.

LOSS-ON-IGNITION OF ORGANIC AND CARBONATE CARBON

Weight percent loss-on-ignition for the determination of organic and carbonate carbon was done on the same 4-cm intervals as the pollen samples at Buttonbush Bog. The original sediment samples from Round Spring Shelter and Gooseneck Site were split through a Carpo riffle box splitter according to Shackley (1975), until a representative sample of approximately 3 g was attained. Initial wet weights of samples from Buttonbush Bog used for analysis were approximately 1 g.

The samples were analyzed according to Dean's (1974) loss-on-ignition technique. Samples were dried at 90-100°C to determine dry weight. Percent loss-on-ignition was determined by combustion at 550°C for one hour for organic carbon and at 1,000°C for one hour for carbonate carbon content. The weight loss of the sample between 550-1,000°C is the amount of CO₂ evolved from carbonate minerals (Dean, 1974). Therefore, the percent CO₂ loss was divided by 0.44, the fraction of CO₂ in CaCO₃. (See Appendix III.D for the complete procedure.)

The following equations were used to calculate weight percents:

$$\text{Weight \% Organic Carbon} = \frac{\text{Total Organic Carbon Lost}}{\text{Dry Sample Weight}} \times 100$$

$$\text{Weight \% CO}_2 \text{ Lost} = \frac{\text{Total \% CO}_2 \text{ Lost}}{\text{Dry Sample Weight}}$$

$$\text{Weight \% Carbonate} = \frac{\text{Weight \% CO}_2 \text{ Lost}}{\text{Fraction of CO}_2 \text{ in CaCO}_3 (0.44)}$$

Eight Buttonbush Bog samples (148, 152, 156, 160, 164, 168, 172, and 176 cm below surface) were duplicated to compare the results of the loss-on-ignition technique. Three of the original samples (160, 164, 168 cm) were spilled during analysis and lost. The duplicates were used for the interpretation. Comparison of the five remaining duplications showed less than five percent variation in the weight percent organic carbon lost. Less than one percent variation occurred for weight percent carbonate lost. Original copies of the data obtained from loss-on-ignition analysis are on permanent file at the Archaeometry Laboratory, UMD.

CLASTIC SEDIMENT ANALYSIS

Clastic sediment analysis for the determination of sand, silt, and clay weight percentages was performed on the sediment samples from Round Spring Shelter and Gooseneck Site. The original sediment samples were split through a Carpo riffle box splitter according to Shackley (1975), until a representative sample of 7.5 g was attained.

The samples were analyzed using a combination wet sieve and pipette method. Samples were air dried and weighed to four decimal places on a Mettler balance to determine initial dry weight. Sand-sized grains were separated by wet sieving through a 4.0 phi screen. The silt and clay fraction was then transferred to a 1000-ml settling cylinder, 20 ml of $\text{Na}(\text{PO}_3)_6$ (100 ml $\text{Na}(\text{PO}_3)_6$ / 1000 ml H_2O) dispersant was added, and the sample fluid increased to 1000 ml using distilled water. The samples were then agitated for one minute and allowed to stand. At 20 seconds, 20 ml of sample fluid was withdrawn at a depth of 20 cm using a pipette. This sample was dried and weighed to determine the total fine fraction (silt and clay). One hour and 51 minutes after settling began, a second aliquot was withdrawn and then dried to determine the clay fraction of the sample. The complete procedure is shown in Appendix III.E.

Percentages of sand, silt, and clay were determined using the following equations:

Sand fraction:

$$\frac{\text{Sieved sample weight}}{\text{Original dry sample weight}} \times 100 = \% \text{ sand}$$

$$100 \% - \% \text{ sand} = \% \text{ silt and clay (actual)}$$

Silt and clay fraction (4.0 ϕ):

$$\text{Sample weight} \times 50 = \text{calculated sample weight}$$

$$\frac{\text{Calculated sample weight}}{\text{Original dry sample weight}} \times 100 = \% \text{ silt and clay (empirical)}$$

Clay fraction (8.0 ϕ):

$$\text{Sample weight} \times 50 = \text{calculated sample weight}$$

$$\frac{\text{Calculated sample weight}}{\text{Original dry sample weight}} \times 100 = \% \text{ clay (empirical)}$$

$$\% \text{ Silt and clay (empirical)} - \% \text{ clay (empirical)} = \% \text{ silt (empirical)}$$

Corrected values for true percentages were determined using the following equations:

$$\% \text{ Clay (actual)} = \frac{ac}{b}$$

$$a = \% \text{ Silt and clay (actual)}$$

$$b = \% \text{ Silt and clay (empirical)}$$

$$c = \% \text{ Clay (empirical)}$$

$$\% \text{ Silt (actual)} = \% \text{ Silt and clay (actual)} - \% \text{ clay (actual)}$$

For complete details of the mathematical calculations see Appendix III.E.

COLLECTION OF CONTEMPORARY VEGETATION

For the purpose of obtaining a comparative reference phytolith collection of plants now growing at the study sites, 150 plant samples were collected. In addition to providing a phytolith reference collection, flowering samples provided material for reference pollen slides, and plants from each site provide an indication of the contemporary vegetation.

Contemporary plant material was collected at Round Spring Site 23SH19, Gooseneck Site 23CT54, Lewis Lake, and Ozark Sink Pond in the study locality and at a cypress swamp in Butler County during July and August 1981. Plants were collected at Buttonbush Bog and one sample was collected along Highway 60, 5 km west of Van Buren in August 1981. Appendix II.A-E lists plants collected by site. The list is meant to provide an indication of contemporary vegetation at the sites but should not be considered exhaustive.

Three complete samples of each plant were collected if possible. Bark, leaves, seeds, flowers, branches, other diagnostic features, and roots if possible were collected from large trees and shrubs. Each plant type was assigned a collection number and pertinent information regarding the plants was recorded on a plant data sheet. The plants were enclosed in newsprint folders and placed between cardboard blotters. A multilayer of plant samples and blotters was then placed between press frames, the entire unit was compressed and then tied.

The plants were then dried. Drying proceeded slowly because of high humidity: newspapers were changed daily until plants were dry. Final drying was accomplished by placing the samples in a low-temperature drying oven at the Olga Lakela Herbarium, University of Minnesota, Duluth. The dried plants were then stored until identification could be undertaken.

PLANT IDENTIFICATION

The contemporary vegetation was identified using the keys in Steyermark (1963), Sargent (1965), Peterson and McKenny (1968), Morley (1969), Britton and Brown (1970), Fernald (1970), and Hitchcock (1971); by referring to pictures and texts in United States Department of Agriculture

(1971), Denison (1978), Phillips (1979), and Wylie and Gass (1980); and by comparison to the plant reference collection at the Olga Lakela Herbarium, University of Minnesota, Duluth. Initial identifications were made by the author if possible and verified by Dr. Paul H. Monson, curator of the Olga Lakela Herbarium, University of Minnesota, Duluth, who identified several plants that could not be identified by the author.

Identifications were made to the lowest taxonomic level possible. The degree of certainty for some taxa is indicated by the use of "cf." or "?". An identification preceded by "cf." was uncertain because of poor preservation, immature specimens, or ill-defined morphology. A "?" before an identification indicates the specimen was too immature to identify or the key characteristics needed for identification (such as seeds, flowers, or bracts) were not present.

Eleven of 150 plant samples collected could not be identified to the family level, and thus were not used for analysis. All nomenclature follows *Flora of Missouri* (Steiermark, 1963), except for the family designation, Corylaceae, which is replaced by Betulaceae. Voucher specimens were made of all specimens analyzed in this study if, after phytolith extraction, enough material was left to do so. The specimens are on file in the Olga Lakela Herbarium, University of Minnesota, Duluth.

Contemporary Vegetation Reference Samples

One hundred and thirty-seven modern plant samples (Appendix IV) were analyzed to determine whether they contained phytoliths, and if so, which morphological types. Before phytolith extraction was undertaken, the plants were subdivided into specific plant parts to be processed. If material was available, four major subdivisions were processed: leaf, stem, inflorescence, and root. Plant parts were further divided if it was deemed necessary; for example, the leaf blade and leaf sheath were separated in the grasses. A total of 525 individual plant parts (Appendix IV) was analyzed. For each plant part, a 0.25-g sample was processed for phytoliths, according to a modified Rovner (1971, 1972) technique using Schulze solution – 3 parts nitric acid (HNO_3) to 1 part saturated potassium chlorate (KClO_3) – to digest the plant material. (See Appendix III.H

for the complete phytolith extraction procedure.) Following extraction, the phytolith samples were stored in 95% ethanol for later analysis.

PHYTOLITH IDENTIFICATION AND COUNTING TECHNIQUES

Processed phytolith samples were mounted in Permount on glass slides using 22 x 30 mm cover slips. For the sediment samples that have been studied in detail at this time (Gooseneck Site 23CT54), a minimum number of 200 phytoliths was counted, except for sample 6 in which only 190 phytoliths were counted. The phytolith sum includes five categories of phytoliths: long trapezoid, short trapezoid, saddle, dumbbell, and cross. Other silica bodies counted but excluded from the phytolith sum are silicified cells of trichome cones, stomata, bulliform cells, epidermal cells, rods, and rectangular/square cells. Unidentifiable silica bodies counted but not included in the phytolith sum were divided into three categories: weathered, tilted, and unknown phytoliths. The above-mentioned types are based on phytoliths derived from grasses and do not take into consideration morphological phytolith types produced by other plant families known to contain phytoliths.

PHYTOLITH ANALYTICAL METHODS

Buttonbush Bog

The core was systematically sampled at the same 4-cm intervals used for pollen analysis and was processed for phytoliths using a technique developed at the Archaeometry Laboratory, University of Minnesota, Duluth. The samples were burned at low temperature (350 °C) for one week to free the phytoliths from the peat material. This part of the procedure is adapted from Dean's (1974) loss-on-ignition of organic carbon technique. The samples were then processed using a modification of the pollen extraction technique (KOH and acetolysis) of Faegri and Iverson (1975) to remove any remaining organics. (See Appendix III.F for the complete phytolith processing procedure.) The samples were then stored in 95% ethanol for later analysis.

Round Spring Shelter, Round Spring Site 23SH19

The sediment samples from Round Spring Shelter were dried at room temperature and split through a Carpco riffle box splitter according to Shackley (1975), until a representative sample of 2-3 grams was attained. The samples were processed using a modified Rovner (1971) technique (tetrabromoethane 2.3 specific gravity separation) to separate the phytoliths from the sediment. The phytolith samples were then stored in 95% ethanol for later analysis. (See Appendix III.G for the complete processing procedure.)

Gooseneck Site 23CT54

Phytolith extraction for the sediment samples was undertaken in the same manner described for Round Spring Shelter. (For a detailed description of the phytolith extraction procedure, see Appendix III.G.)

All phytolith slides were initially scanned at a magnification of 125x on the Leitz Ortholux microscope used for pollen counting. Initial scanning was undertaken to determine the presence or absence and relative abundance of phytoliths on the slide.

The detailed counts of the Gooseneck Site 23CT54 were done on a Zeiss microscope equipped with 2.5x, 10x, 25x, and 40x objectives. The microscope is equipped with 8x oculars, an OPTO-VAR magnification changer (1.25x, 1.6x, and 2x), and Nomarski differential interference contrast system. Phytolith identifications were made using the 1.25x OPTOVAR and the 40x objective, in conjunction with the Nomarski system. All of the Gooseneck Site 23CT54 phytolith counts were done by A.L. Ollendorf, Archaeometry Laboratory, UMD.

THE PHYTOLITH KEY

The phytolith key used for the detailed counts follows a key developed by S.C Mulholland (1984a), Archaeometry Laboratory, UMD and is presented in Table 7. The first four phytolith categories described were included in the phytolith sum. Cross and dumbbell, listed together as the first category, were separated for this study. Categories 5-9 were counted but excluded from the

phytolith sum. Figures 19-23 are photographs of phytoliths representing the five major categories used in this study. All photographs are from S.C. Mulholland (1984b, in press).

TABLE 7: GRASS CLASSIFICATION SCHEME - GENERAL CATEGORIES

I.

- 1) Dumbbell - Planar View: Two lobes separated by a distinct shaft.
Cross - Planar View: Four roughly equal lobes separated by four equal indentations.
- 2) *Saddle - Planar View: Two convex edges separated by two concave edges.
- 3) Long Trapezoid - Planar View: Rectangular or oblong body, often sinuous.
Side View: Trapezoidal.
- 4) Short Trapezoid - Planar View: Circular or oval body.
Side View: Trapezoidal.

II.

- 5) Trichome - Esau, 1977, Anatomy of Seed Plants (2nd edition):
"An outgrowth from the epidermis. Trichomes vary in size and complexity and include hairs, scales and other structures and may be glandular." Includes macrohairs, microhairs and papillae. Usually has a pointed section and a basal section.
 - 6) Long Cell - Thin, roughly rectangular cells, often with projections on the sides. May be silicified together to form a plate, sometimes with stomata or other cells.
 - 7) Bulliform Cell - Esau, 1977: "An enlarged epidermal cell present with other similar cells, in longitudinal rows in leaves of grasses." Keystone or fan-shaped cells of varying shape.
 - 8) Stoma - Esau, 1977: "An opening in the epidermis of leaves and stems bordered by two guard cells and serving in gas exchange." Silicified guard and/or subsidiary cells.
 - 9) Rectangle and Square - Large, blocky cells of rectangular or square shape.
-

*In this study the Saddle-side view was defined as two sides approximately equal in length.



Figure 19. Dumbbell (*Setaria lutescens* (Weigel.) Hubb.). Photo from Mulholland, S.C. (in press).



Figure 20. Cross (*Arundo donax* L.). Photo by Thomson, M., from Mulholland, S.C. (in press).

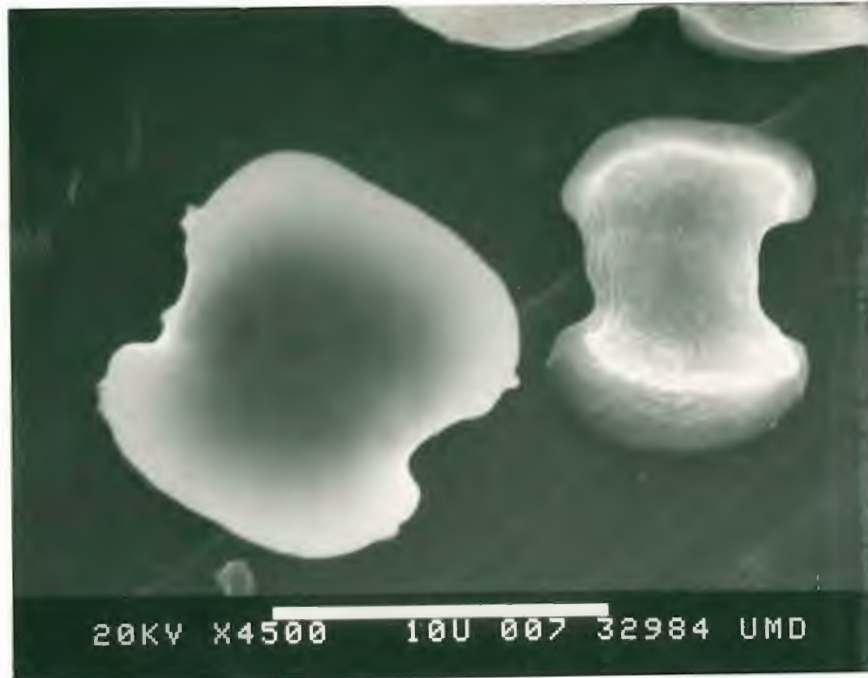


Figure 21. Saddle (*Eragrostis cilianensis* (All.) Lutati). Photo from Mulholland, S.C. (in press).

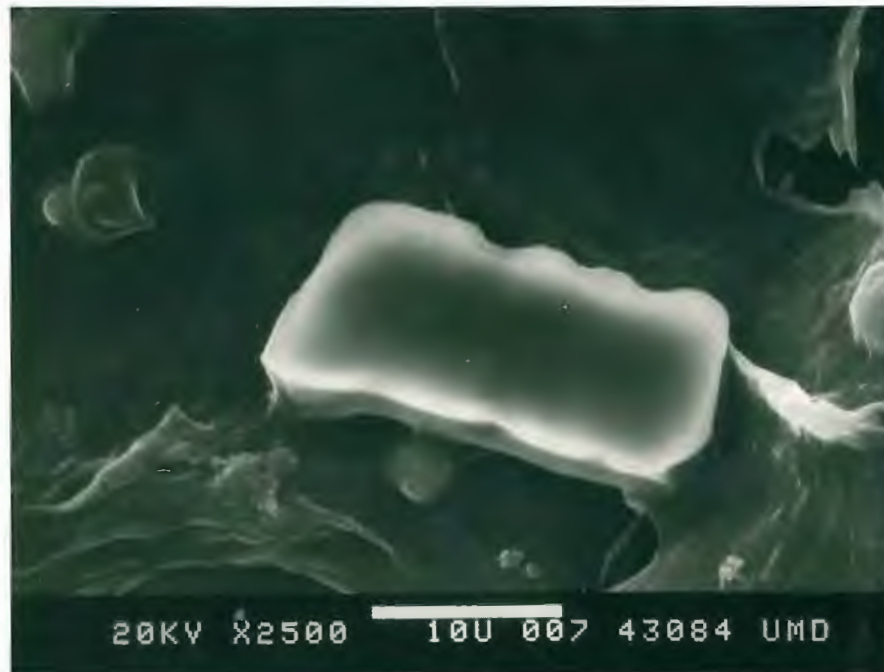


Figure 22. Long Trapezoid (*Bromus inermis* Leyss). Photo from Mulholland, S.C. (in press).

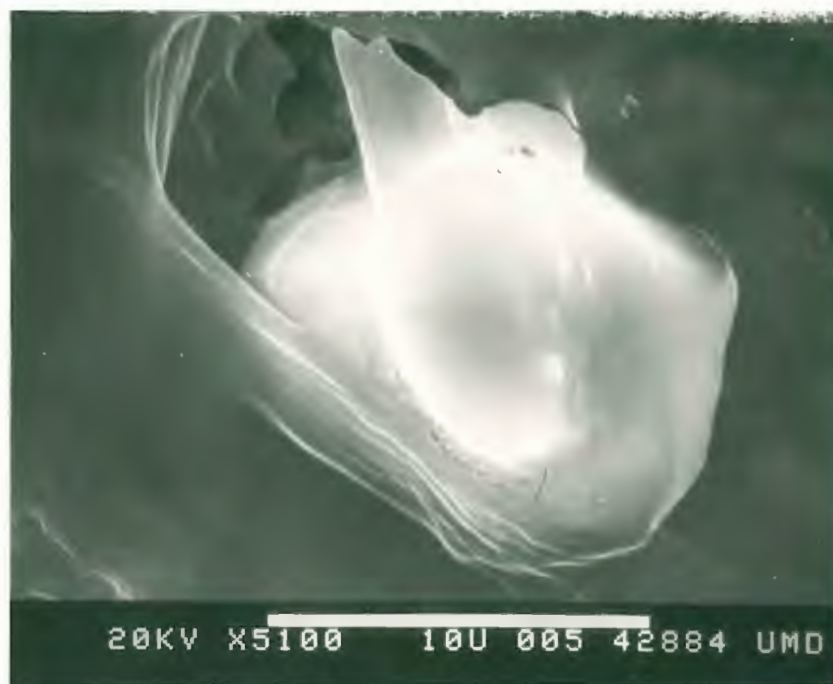


Figure 23. Short Trapezoid (*Phleum pratense* L.). Photo from Mulholland, S.C. (in press).

MODERN POLLEN RAIN

The modern pollen rain percentage diagram (Plate 1) is divided into four parts. The bryophytic polsters are grouped together by site in the top two parts, followed by the three pinch samples, with the regional pollen rain at the bottom. Regional pollen was computed by adding all the pollen counts and calculating the percentages from the total pollen sum. The pollen spectra from the Buttonbush Bog polsters demonstrate the variability of pollen frequency in different samples from the same site. King and Kapp (1963) found similar variation in pollen frequency from single-site samples in eastern Ontario. Counts for the Buttonbush Bog and Ozark Sink Pond bryophytic polsters were combined for each of their respective sites, and percentages were calculated in order to determine a more accurate pollen frequency for both sites.

Pollen frequencies derived from the Buttonbush Bog pinch sample fall within the range of variability observed in frequencies derived from polsters from the same site, indicating that the two methods are comparable. Comparisons of the 95% confidence intervals and point estimates (Plate 2) also indicate no significant difference for many of the samples as a whole, although significant differences occur for some individual taxa, especially *Quercus*.

The modern pollen spectra (Plate 1) are characterized by high percentages of AP (arboreal pollen), 60 to 90%. *Quercus* is the dominant pollen type in the samples, with values of 29 to 67%. It occurs at 51.6% in the regional pollen spectra, with the lowest values at Round Spring and Gooseneck. Pine values range from 5 to 32%; the lowest occur at Ozark Sink Pond. This is to be expected because no pine was observed growing in the vicinity of the site (Appendix II.B). *Fraginus* values are low, less than 7%, the highest value occurring at the site. *Carya* values are approximately 3 to 4% in all the samples except Gooseneck where it reaches 17%, reflecting the local abundance of *Carya* observed at the site. Nonarboreal pollen (NAP) is highest at Round Spring (36.5%), perhaps reflecting disturbance created by park use.

The pollen frequencies for the dominant taxa found at Round Spring are lower than those reported by King (1973) for the same site. This is probably a result of sample variability from a single site, as observed in the Buttonbush Bog samples.

Pinus strobus and *Ephedra* are not known to occur in Missouri and their presence is attributed to long distance transport via wind currents. *Taxodium* does not occur at any of the specific sites sampled but does occur in swamps to the southeast of the sites (Steyermark, 1963). It is probable that *Taxodium* pollen was blown in from the adjacent area.

SEDIMENT STRATIGRAPHY

BUTTONBUSH BOG

The sediment lithology of the Buttonbush Bog core is summarized in Table 8. Sediment composition is shown in Figure 24, along with loss-on-ignition percentages. These results confirm the overall characteristics of the sediments presented in Table 8. Preliminary analysis of macrofossil residue from pollen extraction shows that insect parts and charcoal fragments occur consistently throughout the core.

ROUND SPRING SHELTER, ROUND SPRING SITE 23SH19

The sediment from Round Spring Shelter is characterized by high carbonate content (58-72%). Organic carbon content ranges between 5.8 and 22.3%. The highest values occur above 26 cm, and are approximately twice as large or larger than the values occurring below 26 cm (Figure 25).

Clastic sediment analysis revealed that the upper 34 cm is predominantly sand-sized material (64-79%), with clay the least abundant (less than 11%), and silt making up the remainder. Below 34 cm, sand and silt occur in approximately equal quantities, silt having the highest value except at the 73-77 cm interval where sand has the greatest value. Clay percentages are low (10% or less) with all but two samples below 6% (Fig. 25).

During coring, a stone (Fig. 26) measuring approximately 10 x 6 x 5 cm and covered with a charcoal film was encountered at 65 cm. The stone has been identified by Dr. S. Aschenbrenner, archaeologist, University of Minnesota, Duluth, as a probable hearth stone. Pollen analysis was undertaken on charcoal scrapings from the stone but no pollen was present.

Munsell colors were determined under artificial light for the Round Spring Shelter samples while they were moist. The observed Munsell colors and their numbers are shown in Table 9.

TABLE 8. SEDIMENT LITHOLOGY AT BUTTONBUSH BOG

Depth* in cm	Type	Munsell ² Color	Munsell ² Number	Lower Contact	Remarks
0 - 121	Fibrous peat	Dark reddish brown	5 YR 2.5/2	Gradational	Recognizable twigs present
121 - 156	Fibrous peat	Dark reddish brown	5 YR 3/2	Sharp	Recognizable herbaceous stems and woody roots occur
156 - 160	Amorphous peat	Black	5 YR 2.5/1	Sharp	Carbonized flecks present
160 - 193	Amorphous peat	Dark reddish brown	5 YR 3/2	Gradational	No woody plant parts present
193 - 224	Amorphous peat	Dark reddish brown	5 YR 3/3	Gradational	Wood fragments up to 1 cm long occur between 198 and 203 cm
224 - 253	Clayey peat	Dark reddish brown	5 YR 3/3	Gradational	Woody stems and roots occur between 235 and 246 cm
253 - 276	Clayey peat	Dark reddish brown	5 YR 3/4	Gradational	3-cm woody stem at 276 cm
276 - 280	Peaty clay	Dark reddish gray	5 YR 4/2	Sharp	Organic components decrease with depth
280 - 302	Organic clay	Dark reddish gray	5 YR 4/2	Not observed	Organics in the form of fine particulates

*Depths are measured from the surface of the peat at the time of coring.

²Munsell colors were determined on wet sediment under artificial light.

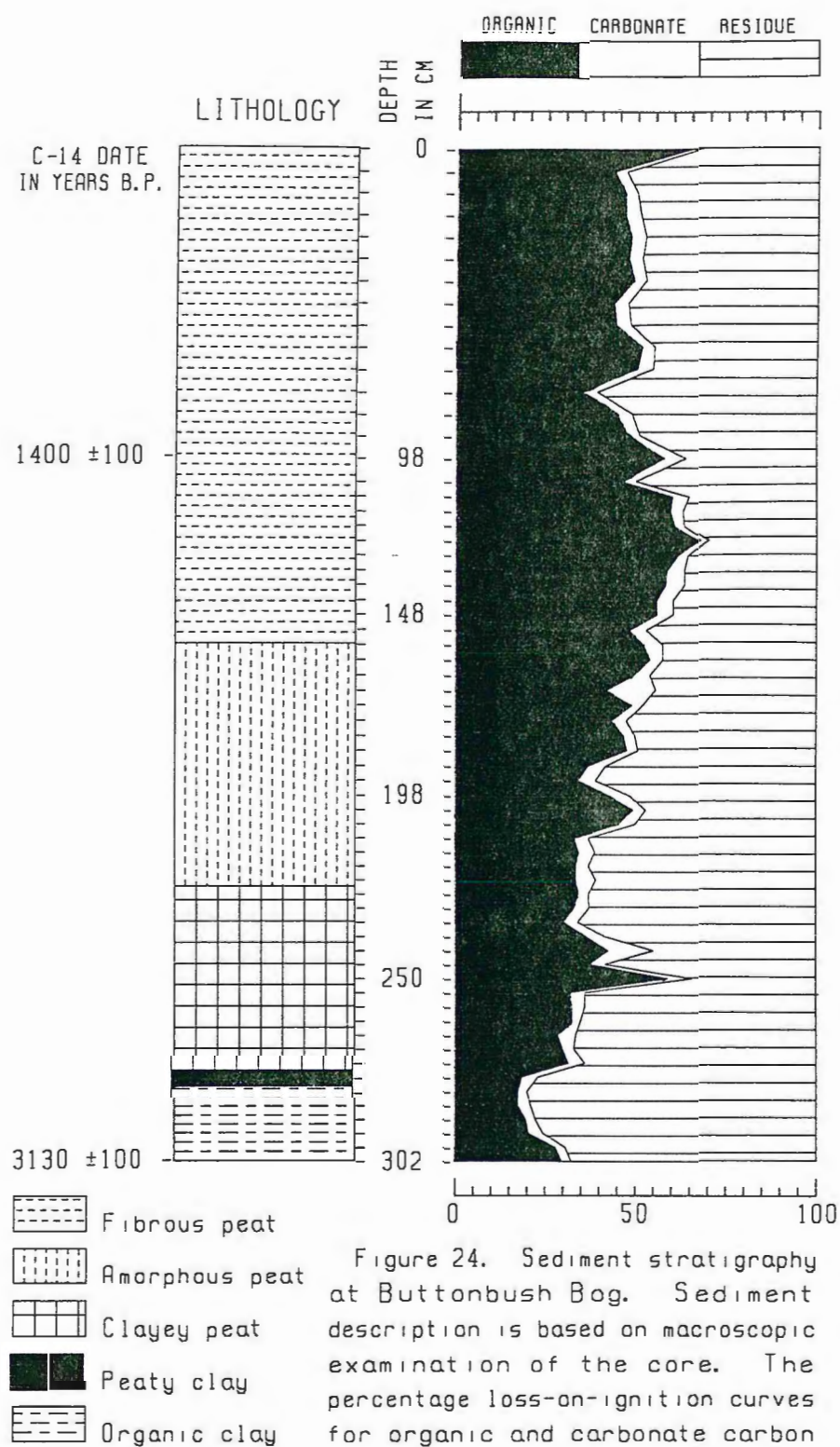


Figure 24. Sediment stratigraphy at Buttonbush Bog. Sediment description is based on macroscopic examination of the core. The percentage loss-on-ignition curves for organic and carbonate carbon are also shown.

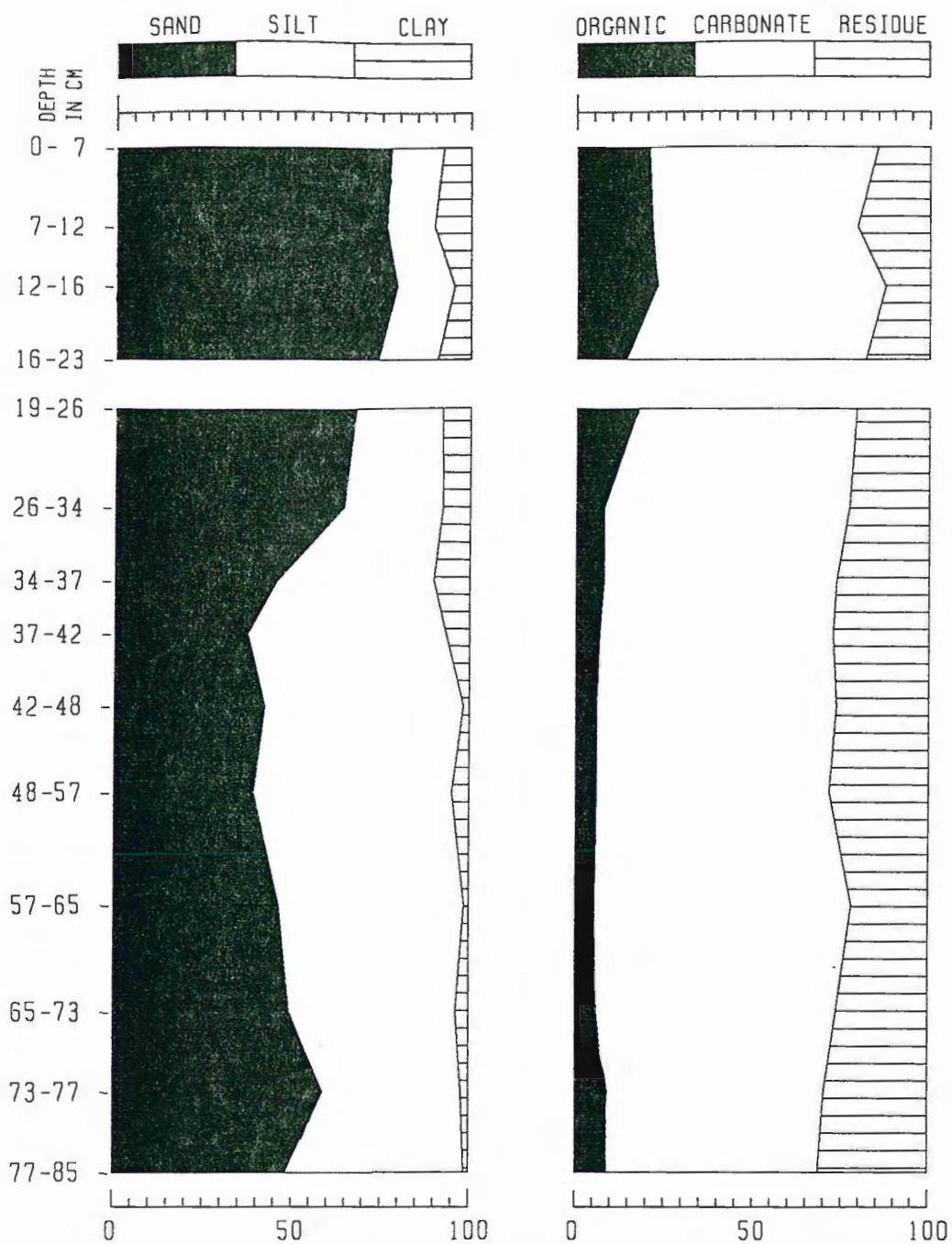


Figure 25. Sediment stratigraphy at Round Spring Shelter, Round Spring Site 23SH19. The percentage curves of sand, silt, and clay and loss-on-ignition of organic and carbonate carbon are plotted against depth. Note that the point representing the sample is plotted at the center of the sample interval.

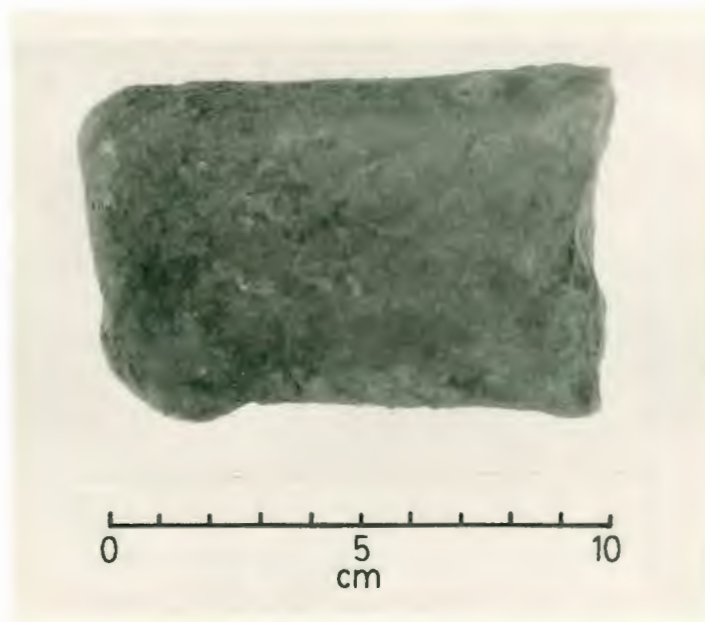


Figure 26. Photograph of the probable hearth stone from the Round Spring Shelter core. Note the darker charcoal film in the lower left area of the stone. Photo property of author.

TABLE 9. MUNSELL COLOR DESCRIPTIONS FOR ROUND SPRING SHELTER

Depth in cm*	Color#	Number
0 - 7	5 YR 3/3	Dark reddish brown
7 - 12	5 YR 3/2	Dark reddish brown
12 - 16	5 YR 3/3	Dark reddish brown
16 - 23	5 YR 3/4	Dark reddish brown
19 - 26	5 YR 4/6	Yellowish red
26 - 34	5 YR 5/8	Yellowish red
34 - 37	5 YR 4/6	Yellowish red
37 - 42	5 YR 5/6	Yellowish red
42 - 48	5 YR 5/6	Yellowish red
48 - 57	5 YR 5/8	Yellowish red
57 - 65	5 YR 5/4	Reddish brown
65 - 73	5 YR 5/8	Yellowish brown
73 - 77	5 YR 5/6	Yellowish brown
77 - 85	5 YR 5/6	Yellowish brown

*Depth measured from the surface of the shelter floor at the time of coring.

#Munsell colors were determined on moist sediment under artificial light.

GOOSENECK SITE 23CT54

Lynott (1981, written commun.) describes the sediment at Gooseneck Site as having a dark upper sandy loam horizon overlying a lighter more compact subsoil. The A-P horizon is thinnest on top of the terrace and thickest at the edge. The deposit at the terrace's edge has been modified by human activity, and appears to be a midden zone. The soil pH at the site is generally 5.0-6.0. Figure 27 is a typical soil profile from the terrace edge.

The eight sediment samples were assigned arbitrary numbers (1-8) for convenience and will be referred to as such. Table 10 shows the arbitrary number, test unit coordinates, sample depth, level provenience, and Munsell soil color and number for each sample.

Four sediment samples (2, 3, 6, 7) were collected from the midden area along the terrace edge. Samples 4 and 8 are from what appears to be a house feature (Lynott, 1981, written commun.). Samples 1 and 5 were collected from the A-P and subsoil horizons. The soil profiles for the test units from which the samples were collected are shown in Figure 28.

The sediments from Gooseneck Site are 56-70% sand, 25-41% silt, and the rest clay (Fig. 29). As a result of varying sample intervals, provenience comparisons were not attempted.

Loss-on-ignition results show very low values for carbonates, less than 1.2% (Fig. 29). Values this low may indicate OH ions removed from clays between 550-1000°C (Dean, 1974). Organic percentages are also low, less than 2% except for sample 4, which is 4.9% (Fig. 29). Although this value is low, the greater abundance of organic carbon may be significant because it comes from an archaeological house feature and may reflect organic remains of the house.

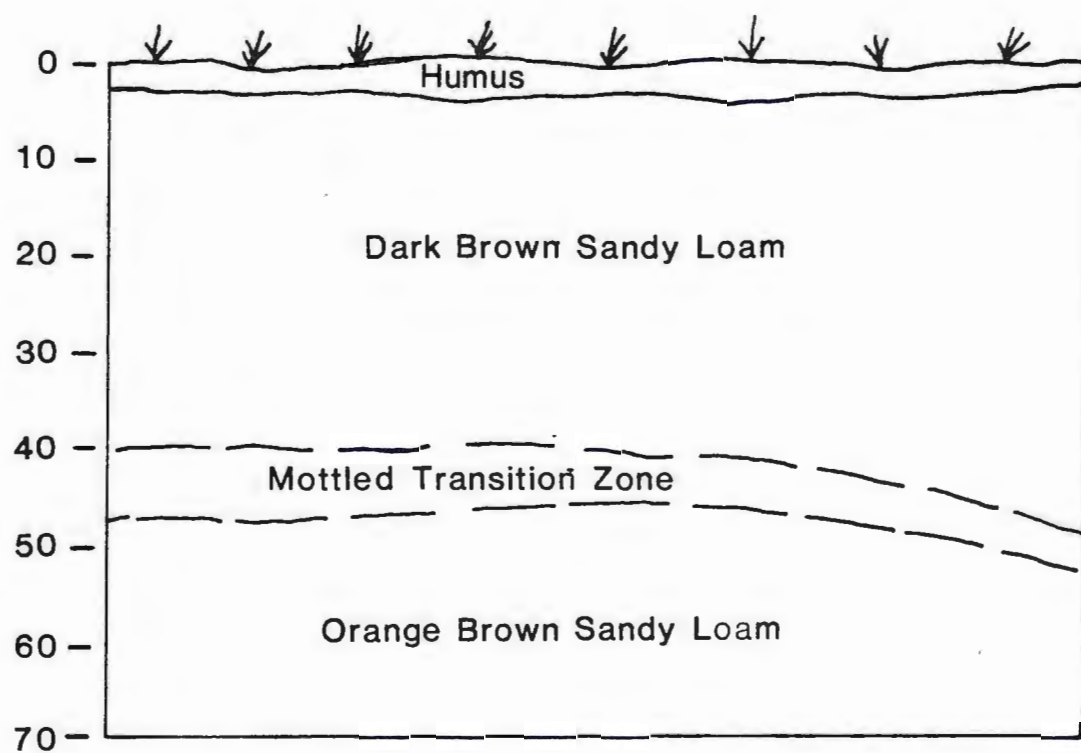


Figure 27. Typical soil profile of the terrace edge, Gooseneck Site 23CT54. Redrawn from National Park Service, 1980.

TABLE 10. SAMPLE LOCATIONS AND MUNSELL COLORS FOR GOOSENECK SITE 23CT54

Arb ¹ no.	Test unit ² coordinates	Depth ³ in cm	Level provenience	Munsell ⁴ color	Munsell ⁴ number
1.	22N/4-5E	0-25	A-P Horizon	Very pale brown	10 YR 7/4
5.	22N/4-5E	25-30	Subsoil	Yellow	10 YR 8/8
2.	22-23N/22-23E	20-30	Midden	Yellowish brown	10 YR 5/8
6.	22-23N/22-23E	30-40 (>30)	Submidden	Very pale brown	10 YR 7/4
3.	22-23N/24-25E	20-40	Midden	Yellowish brown	10 YR 5/6
7.	22-23N/24-25E	40-50 (>40)	Submidden	Very pale brown	10 YR 7/4
4.	31-32N/12-13E	10-20	House feature	Yellowish brown	10 YR 5/8
8.	31-32N/12-13E	20-30	House feature	Light yellowish brown	10 YR 6/4

¹Arb no. = Arbitrary number assigned to each sample.

²Coordinates are from assigned datum at site (Fig. 18).

³Depth below surface.

⁴Munsell colors were determined on dry sediment under artificial light.

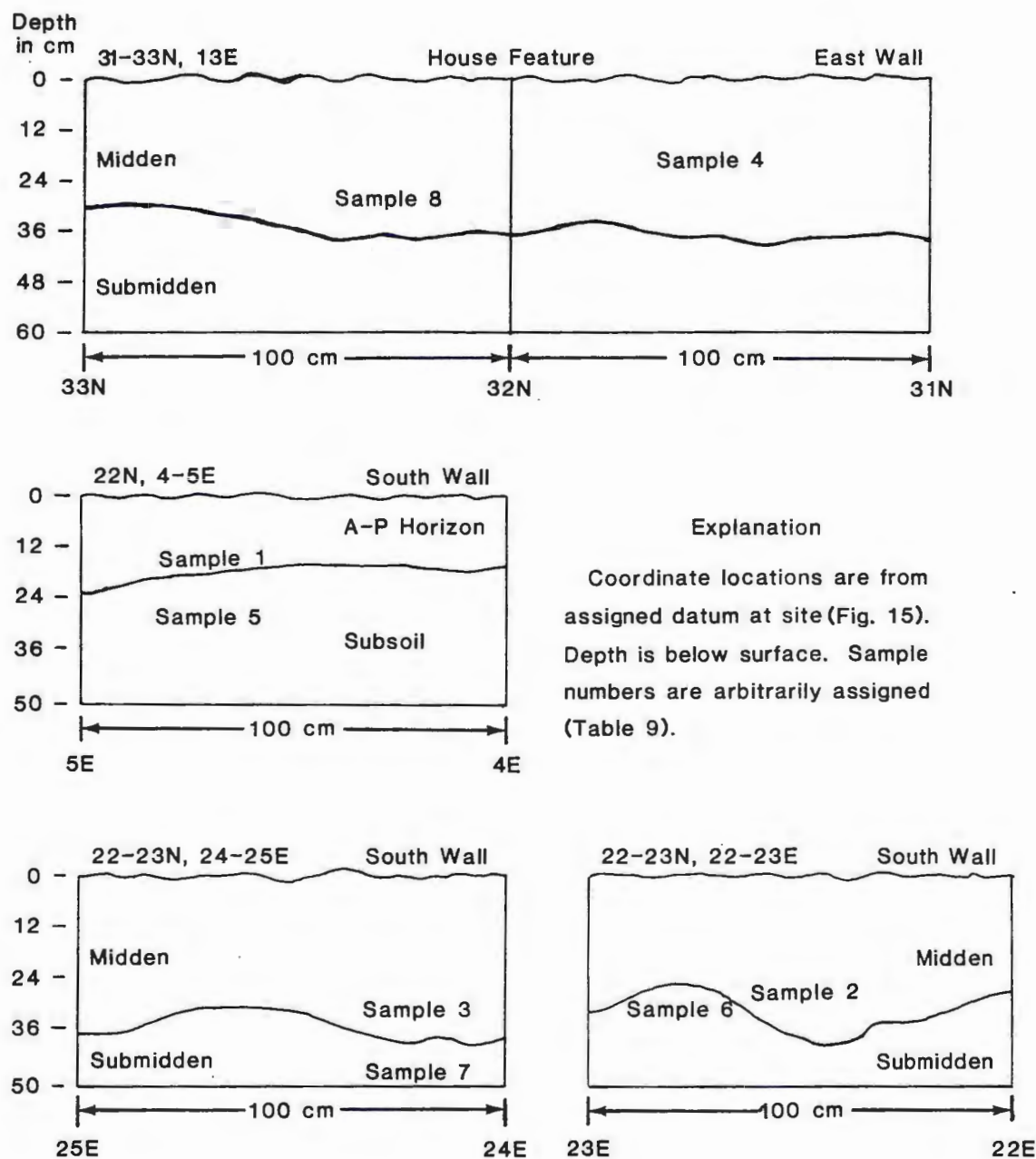


Figure 28. Gooseneck Site 23CT54 test unit soil profiles for samples collected. Redrawn from National Park Service, 1980.

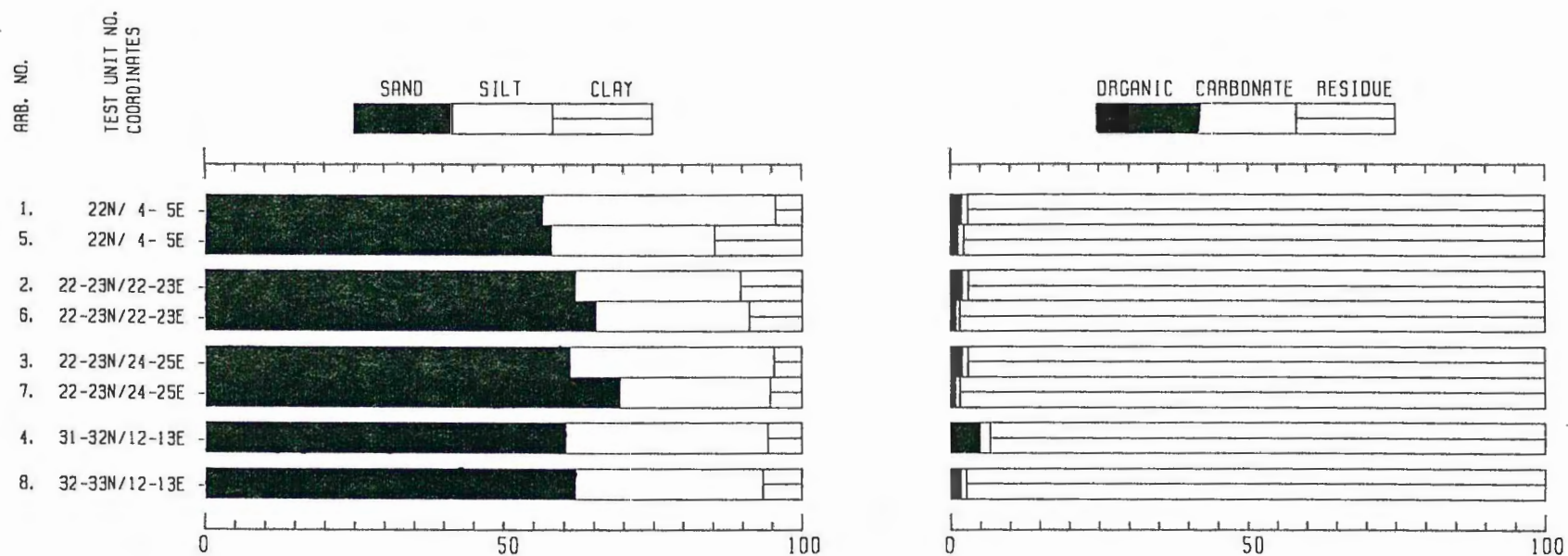


Figure 29. Sediment stratigraphy at Gooseneck Site 23CT54. The percentage curves of sand, silt, and clay and loss-on-ignition of organic and carbonate carbon are plotted against level provenience. (Arb. no. = arbitrary sample number, see Table 10).

RADIOCARBON DATES AND SEDIMENTATION RATES

BUTTONBUSH BOG

Two peat samples and an organic-rich clay from the Buttonbush Bog core were submitted for radiocarbon dating; their depths, radiocarbon ages (years B.P.), and laboratory numbers are:

52 - 56 cm 1400 ± 100 years B.P. UCR - 1535

98 - 102 cm 980 ± 70 years B.P. UCR - 1536

298 - 302 cm 3130 ± 100 years B.P. UCR - 1537

Subsequent examination of the core indicates that the inverted date of 980 ± 70 years B.P. probably resulted from incorporation of younger peat material in the core barrel during the second penetration. The texture of the peat from 98-108 cm was loosely compacted and less firm than peat from below 108 cm, and this section is assumed to mark the extent of the younger incorporated peat. Peat from this section was not used for pollen analysis or sedimentation rates.

During extrusion, the uppermost section of the core underwent the greatest compaction, creating a significant difference between penetration depth and extruded length (a reduction of 40 cm). The sample intervals have been adjusted to represent actual penetration depth.

The slope of the line produced by plotting the two dates against depth indicates the apparent sedimentation rate for the Buttonbush Bog core (Fig. 30). Between 3130 and 1400 years B.P., peat and organic clay accumulated at the rate of 0.12 cm/yr (8.3 yr/cm) and from 1400 years B.P. until present at 0.07 cm/yr (14.3 yr/cm). During the last 1400 years, sediment accumulation slowed approximately 42%, representing about 32% of the core retrieved from Buttonbush Bog.

It is assumed that the surface of the bog represents the present or time zero. If the sediment had continued to accumulate at the same rate as inferred from the radiocarbon dates, the bog surface would be 655 years old (Fig. 30: dashed line). This would indicate either that no peat formed in the last 655 years or that the upper peat has been removed by some means. However, several factors indicate that the bog surface represents the present and is still actively accumulating peat. As mentioned above (See Specific Site Descriptions), until 1975 the bog was covered by shallow

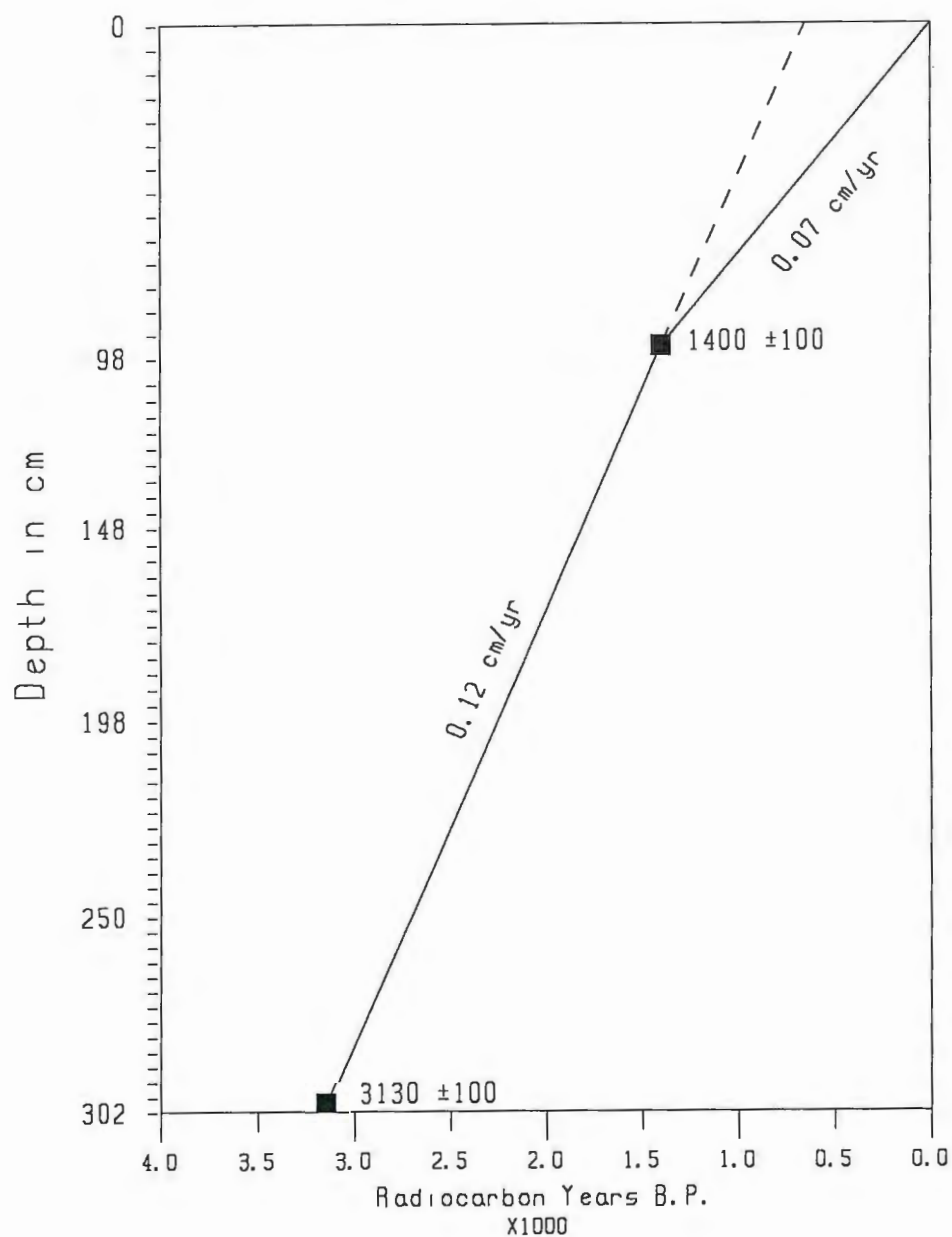


Figure 30. Radiocarbon dates from the Buttonbush Bog Core plotted against depth. The inferred sediment accumulation rates were calculated from adjacent pairs of dates.

water, suggesting that peat accumulation has recently reached the upper boundary of the water table. Except for a small area in the center, the bog is currently covered with vegetation and shows no sign of surface erosion. Furthermore, a small *Ambrosia* rise (Plate 3) in the upper few levels of the pollen diagram, usually found in conjunction with land clearance in the midwest (Huber, 1980, 1985; King 1981; Van Zant, 1976; 1979), indicates that peat was still accumulating when the area was first settled by pioneers about 1820 (Sauer, 1920).

POLLEN CONCENTRATION AND INFLUX RATES

BUTTONBUSH BOG

Pollen concentration for the pollen sum varied from 27,507 to 1,116,991 grains/cm³; the highest value occurred at the clayey peat/peaty clay transition zone (Fig. 31). Concentrations for the total fossil palynomorphs counted range from 66,376 to 2,047,817 grains/cm³. The highest and lowest values correspond to the same sample levels as the pollen sum concentrations.

The pollen concentrations fluctuate greatly throughout the core (Fig. 31). However, the general trend is an increase in pollen concentration from the bottom of the core to the clayey peat/peaty clay transition (270 cm) and then a decrease to 98 cm where concentration drops significantly, reaching its lowest value of 27,507 grains/cm³. From 98 to 70 cm, pollen concentration increases to approximately 200,000 grains/cm³. Then, although fluctuating, concentration continues a general decrease to 28 cm, then increases slightly to the surface. The total fossil palynomorph concentrations (Fig. 31) and the individual taxa that occur continuously throughout the core (Plate 4) reflect the same trend.

The concentration value at the clayey peat/peaty clay transition is probably the result of slow peat accumulation in the initial stages of bog development. King and Allen (1977) recorded a similar situation in the Old Field swamp core. The large drop in concentration shown in both pollen sum and total fossil palynomorph data above 98 cm (Fig. 31) may result from several factors. First, the upper 98 cm of peat had a high water content and was loosely compacted. It may be that this section of peat became more compressed over time and the pollen became more concentrated. Second, the bottom two meters may already have undergone a process similar to that just described. In addition, the degradation of the peat in the lower section probably concentrated the pollen further by reducing the amount of peat material. Third, the sediment accumulation rate is based on the slope of a line between two points and is assumed to be linear, which may or may not be true. If the accumulation of sediment did not follow a linear relationship and instead fluc-

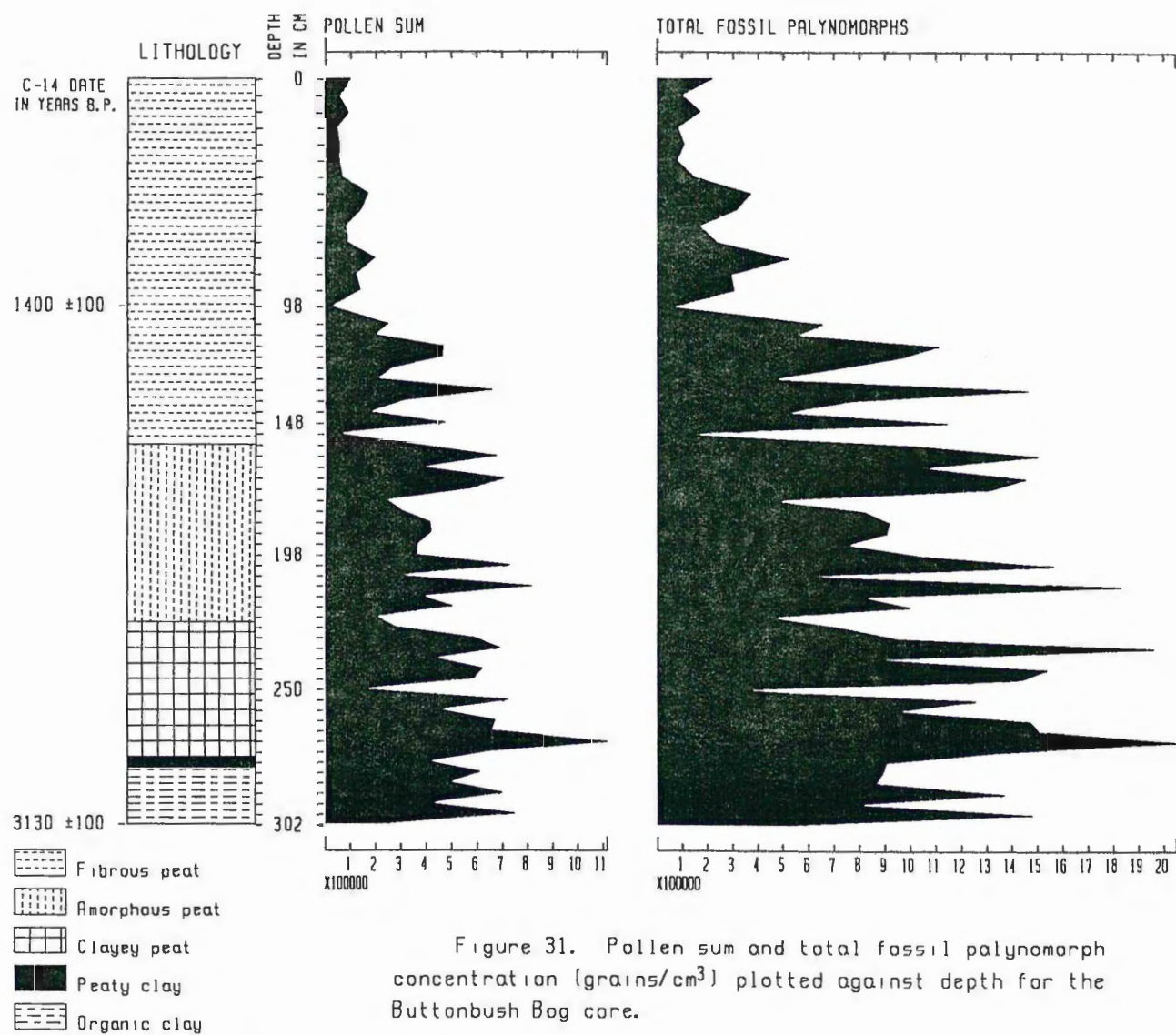


Figure 31. Pollen sum and total fossil palynomorph concentration (grains/cm³) plotted against depth for the Buttonbush Bog core.

tuated throughout the core or assumed a sigmoidal curve, there might have been a much more gradual change in pollen concentration in the core.

Another possibility is that the sample for the the inverted radiocarbon date occurred in this section of the core, and was not used for analysis because it appeared to be younger material incorporated into the core barrel during the second penetration. The material removed may have contained some of the naturally accumulated peat and the deletion of the assumed incorporated younger peat may have created a hiatus in the core. A radiocarbon date from the top of the second section of peat used for analysis may answer this question.

The pollen influx rates for the pollen sum and total palynomorphs are plotted against depth in Figure 32. The pollen influx followed the same trend as pollen concentration (Fig. 31), with the highest and lowest values occurring at the same sample intervals. The pollen influx rates ranged from 1925 to 134,038 grains/cm² for the pollen sum and from 4646 to 245,738 grains/cm² for the total fossil palynomorphs. The large drop in the pollen influx rate after 1400 B.P. may be real but is probably a result of the same factors described above that may have influenced the pollen concentration. The pollen influx rates for individual taxa (Plate 5) also show the same trend but the pollen percentages, except for Gramineae, do not show any significant changes associated with this problem area (Plate 5).

ROUND SPRING SHELTER, ROUND SPRING SITE 23SH19

Pollen concentration for the pollen sum varied from 1980 to 7977 grains/cm³, the highest value occurring in the 57-65 cm interval (Fig. 33). Note that the point representing each sample is plotted at the center of the sample interval in Figure 30. The gap in the diagram (Fig. 33) is at the 4-cm overlap in the sample intervals described above (See Coring Methods and Collection of Archaeological Sediments, Round Spring Shelter). The Round Spring Shelter core has not been radiocarbon dated; therefore, pollen influx rates could not be calculated.

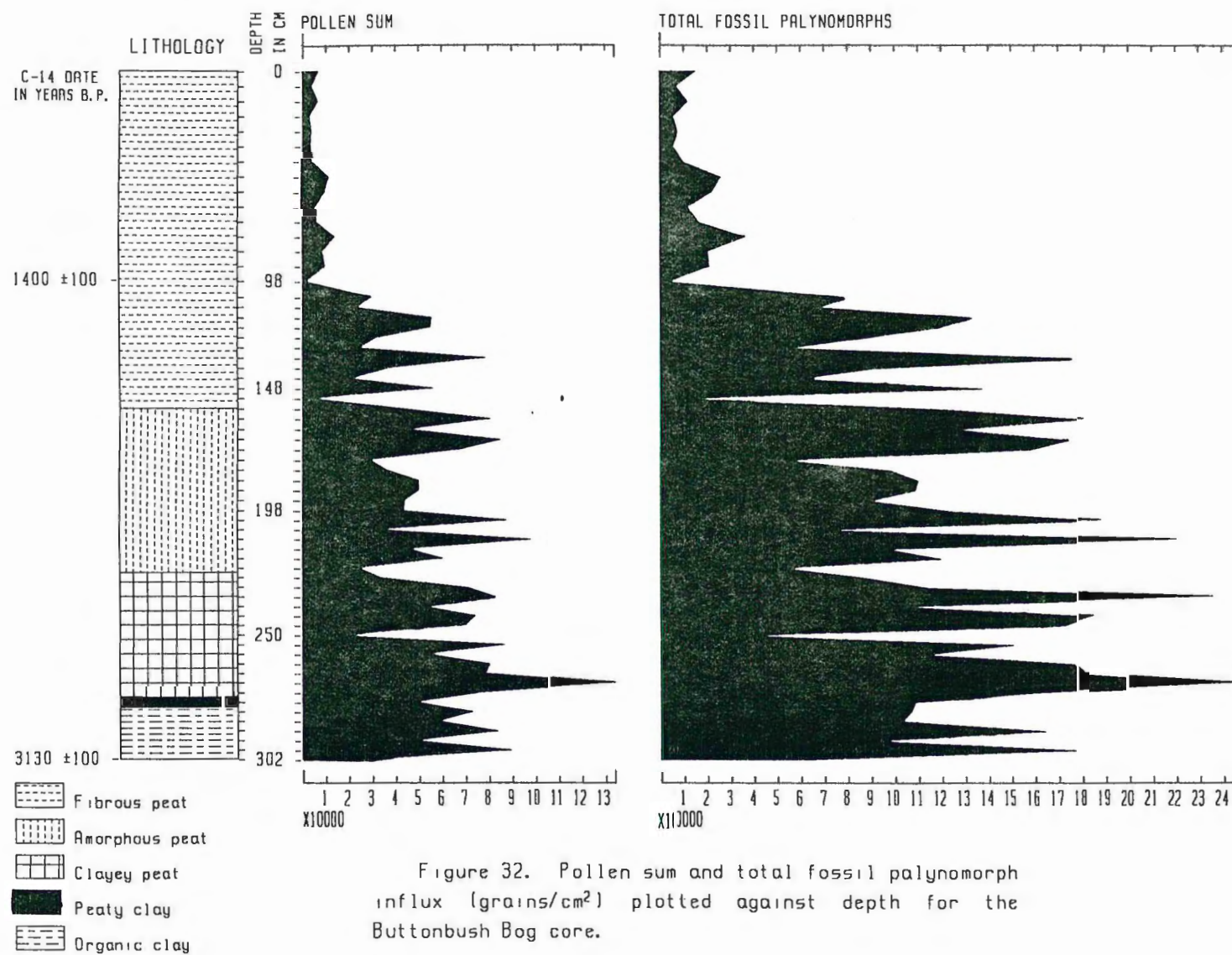


Figure 32. Pollen sum and total fossil palynomorph influx (grains/cm²) plotted against depth for the Buttonbush Bog core.

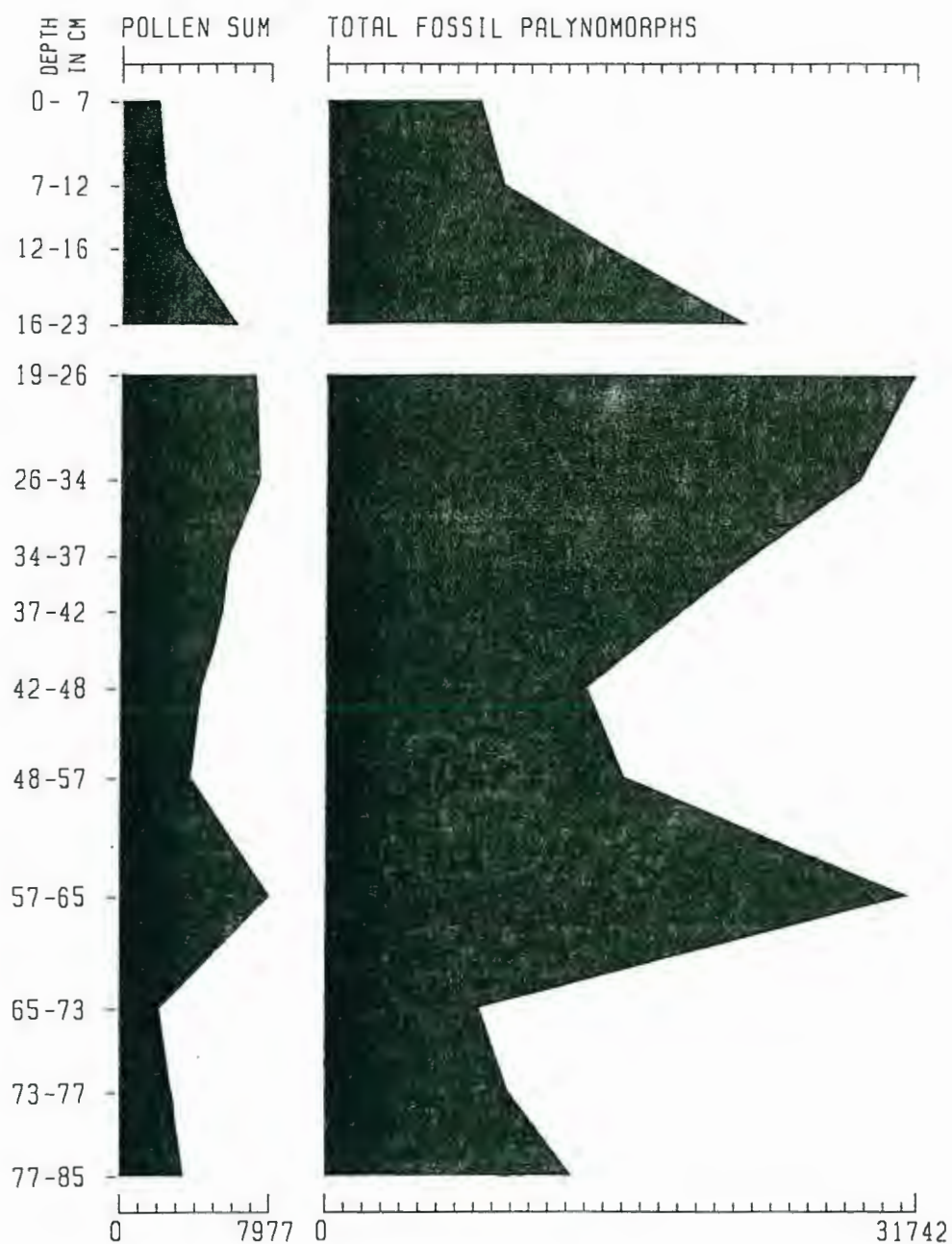


Figure 33. Pollen sum and total fossil palynomorph concentration (grains/cm³) plotted against depth for the Round Spring Shelter core.

Between the 77-85 cm and 65-73 cm sample intervals, the pollen sum concentration decreases gradually from 3369 to 2103 grains/cm³. The concentration then increases significantly to 7977 grains/cm³ in the 57-65 cm interval, and decreases to 3715 grains/cm³ in the 48-57 cm interval. From the 48-57 cm interval, pollen sum concentration increases gradually to the 26-34 cm interval and then decreases gradually to the top sample interval (Fig. 33). The overlapping sample intervals varied by approximately 1000 grains/cm³ (a difference of 14.3%) for the pollen sum between samples.

The total fossil palynomorph concentrations ranged from 8255 to 32,742 grains/cm³, the highest value occurring in the 19-26 cm interval. This corresponds to the lower of the overlapping intervals (Fig. 33).

Total fossil palynomorph concentration (Fig. 33) gradually decreases from 13,225 to 8267 grains/cm³ between the bottom sample (77-85 cm) and the 65-73 cm interval, then increases significantly to 31,265 grains/cm³ in the 57-73 cm interval. Concentration decreases rapidly to 14,081 grains/cm³ in the 42-48 cm sample. From the 42-48 cm interval, concentration decreases sharply from 31,742 to 22,583 grains/cm³, a difference of 28.9%. From the 16-23 cm interval, concentration decreases gradually to 8255 grains/cm³ in the uppermost sample (0-7 cm).

GOOSENECK SITE 23CT54

For convenience, pollen concentration will be discussed using arbitrary sample numbers (test unit coordinates are shown in Fig. 34). At Gooseneck, pollen sum concentration ranges from 2017 to 12,666 grains/cm³ (Fig. 34). Sample 1 overlies sample 5 (Fig. 34), from the A-P and subsoil horizons, respectively. Pollen sum concentration decreases from 2690 to 2017 grains/cm³ between the two soil horizons. Total fossil palynomorphs, on the other hand, increase from 6013 grains/cm³ in the subsoil to 6582 grains/cm³ in the A-P horizon.

Midden sample 2 overlies submidden 6 (Fig. 34) and has pollen sum concentrations increasing from 3297 to 12,666 grains/cm³ from the submidden to the midden level. Total fossil palynomorphs increase in a similar manner from 9322 to 26,481 grains/cm³. Samples 3 and 7 are also from

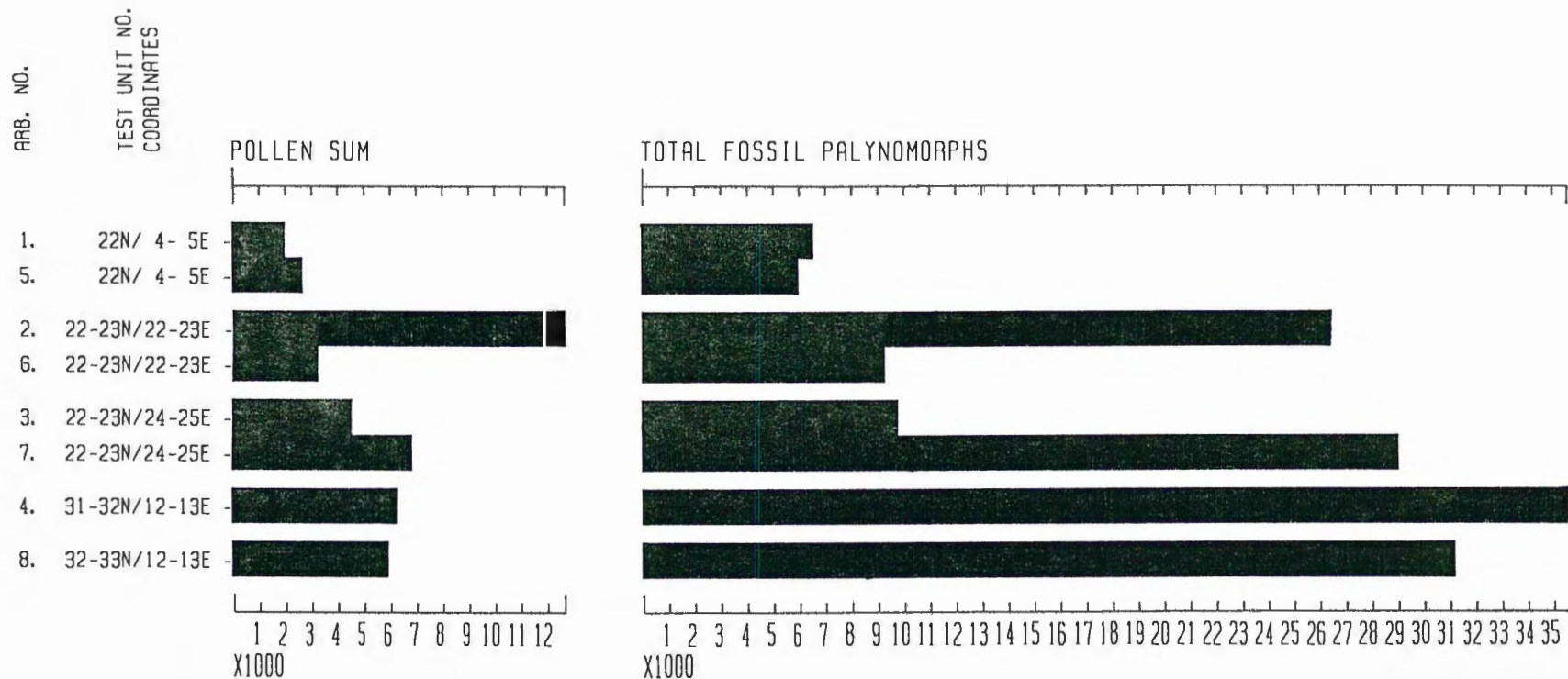


Figure 34. Pollen sum and total fossil palynomorph concentration (grains/cm³) plotted against level provenience for the Gooseneck Site 23CT54 samples. (Arb. no. = arbitrary sample number, see Table 10).

the submidden and midden levels. Pollen sum concentration decreases from 6894 grains/cm³ in submidden sample 7 to 4584 grains/cm³ in midden sample 3, while the concentration of total fossil palynomorphs decreases from 29,065 to 9801 grains/cm³.

Samples 4 and 8 are from adjacent test units but different depths of a house feature (Fig 34). The pollen sum concentration is 6275 grains/cm³ for sample 4 and 5980 grains/cm³ for sample 8. Total fossil palynomorph concentrations are 35,560 grains/cm³ and 31,209 grains/cm³ for samples 4 and 8, respectively.

The differences in pollen sum and total fossil palynomorph concentrations found in the different test units at Gooseneck Site 23CT54 may be a result of human activity and may indicate site specific use. On the other hand, the differences are more likely a result of differential preservation caused by soil oxidation or soil microbes (King and others, 1975).

THE POLLEN DIAGRAMS

BUTTONBUSH BOG

The pollen spectra from the Buttonbush Bog core are dominated throughout by *Quercus*, *Pinus*, and Gramineae (Plate 3) with *Carya* maintaining low but consistent percentages. In all but the three lowermost samples, arboreal pollen (trees and shrubs) comprises more than 60% of the pollen sum. The pollen diagrams are divided into three pollen-assemblage zones. *Pinus* half grains and *Pinus echinata*-type grains are shown in overlay on the percentage, concentration, and influx diagrams (Plates 3,4,5) and are combined on the 95% confidence interval diagram (Plate 6).

Zone 1: *Quercus* Pollen - Assemblage Zone (292-302 cm)

Zone 1 is characterized by 48 to 53% *Quercus*, and less than 15% Gramineae with *Pinus* occurring below 5% (Plate 3). *Carya* occurs with values of 2-3%. *Frazinus* (*F. quadrangulata* and *F. pennsylvanica* *F. americana*) ranges from 0.2 to 4%. *Cephalanthus* and *Salix*, both wet ground shrubs, have values between 1 and 5%. *Ulmus*, *Nyssa*, and *Liquidambar* occur consistently with values of less than 1%. *Populus*, *Platanus*, *Ostrya/Carpinus*, *Acer saccharum*, *Castanea*, *Robinia*, *Betula*, *Morus*, *Juglans nigra*, and *Juglans cinerea* occur occasionally in trace amounts. Shrubs occurring in trace amounts in Zone 1 include *Cornus*, *Rubus*, *Sambucus*, *Vitis*, and *Alnus*.

Juniperus and *Taxodium* both occur in Zone 1. *Taxodium* grains are characterized by an exit papilla (Kapp, 1969) that is not readily apparent when the grains are crushed or broken, making *Taxodium* grains indistinguishable from *Juniperus*. Therefore, only grains exhibiting the exit papilla were identified as *Taxodium*. Although *Taxodium* and *Planera* are both associated with swamps and low wet woodlands, they are not known in the immediate vicinity of Buttonbush Bog today, but do occur in adjacent counties to the east and southeast.

At the time Zone 1 pollen was deposited, *Pinus strobus*, *Picea*, *Tsuga*-type, *Larix*-type, and *Ephedra* pollen probably did not occur in Missouri; nor did *Myrica* and *Taxus* of Zone 2. Their in-

termittent presence throughout the core is attributed to long-distance transport via wind currents. It is also possible that some of this pollen could be derived from older aeolian eroded deposits.

Gramineae and Cyperaceae comprise 70 to 80% of the herbs in this zone (Plate 3). Gramineae and Cyperaceae increase from 8 to 13.6% and 12.1%, respectively. *Equisetum* and *Ambrosia* both have percentage values between 1 and 3%. *Artemisia*, Chenopodiaceae/Amaranthaceae, *Cystopteris bulbifera*, and *Ranunculus*-type occur throughout Zone 1 with values of less than 1%.

One pollen grain found in the 302 cm level count appears to be *Tillandsia usneoides* (Spanish moss) when compared to reference material. *Tillandsia* is not native to Missouri but does occur to the south and southeast. Its occasional occurrence in the core is also attributed to long-distance transport.

The influx rate (Plate 5) for *Pinus* is 492 grains/cm² and for *Quercus* is 14,359 grains/cm² at 3130 years B.P. At this same date *Carya* influx is 845 grains/cm² and Gramineae influx is 2393 grains/cm². There is a sharp increase followed by a decrease in pollen influx for most individual taxa in this zone (Plate 5). Pollen sum influx rates range from 29,600 to 49,550 grains/cm² (Fig. 32) and concentrations range from 246,650 to 747,800 grains/cm³ (Fig. 31) in Zone 1. The pollen concentrations for the individual taxa in Zone 1 are shown in Plate 4.

The low values of *Pinus* in Zone 1 correlate well with the pollen diagram from Old Field swamp (King and Allen, 1977), which shows approximately 5% *Pinus* in sediments younger than 4830 years B.P. The high percentages of *Quercus* are consistent with the modern pollen rain reported in this study (Plate 1) but higher than those reported by King (1973) for sites in Shannon County. The *Quercus* percentages are also higher than those shown on Peterson's (1978) *Quercus* isopoll map, but are within the range of percentages recorded for various sites in Missouri. The *Carya* values compare favorably with those reported by King (1973) and Peterson (1978). Gramineae percentages are higher than those reported by King (1973) and this study (Plate 1) but similar to Peterson's (1978) values. The Gramineae values are only 5% less than those reported by Van Zant (1976, 1979) on the northern edge of the Prairie Peninsula at Lake West Okoboji in northwestern Iowa.

The presence of *Potamogeton* and other aquatics indicates that the sinkhole may have contained open water at this time. *Cephalanthus* and *Salix* are genera usually associated with open water (Steyermark, 1963; King and Allen, 1977). Their occurrence in this zone as well as the clayey basal sediments (Table 8) suggests open water during the deposition of Zone 1. The dominance of oak and the low values of hickory and pine suggest that Zone 1 represents a mixed oak forest with minor components of hickory and pine.

Zone 2: Quercus-Carya-Pinus Pollen-Assemblage Zone (17-292 cm)

In Zone 2, *Quercus* (Plate 3) is still the dominant pollen type, but decreases sharply from 53 to 40%. *Quercus* fluctuates between 26 and 52%, showing a general decline to 34.5 cm followed by a slight increase to Zone 3. *Pinus*, although still low, begins a general increase to 25% at 126 cm, then decreases to 8.7% at 85.5 cm. Above 85.5 cm, *Pinus* increases again to 26.3% to 34.5 cm, then decreases to 17.4% at the top of Zone 2. *Carya*, although fluctuating slightly above or below, occurs consistently with values of 2-4%. *Fraxinus* (*F. quadrangulata* and *F. pennsylvanica*/*F. americana*) occurs consistently with values of less than 5%. *Ulmus*, *Populus*, *Nyssa*, *Liquidambar*, and *Platanus* continue to be relatively consistent minor components of the pollen spectra. *Juglans nigra*, *J. cinerea*, *Ostrya*/*Carpinus*, *Acer saccharum*, *Castanea*, *Robinia*, *Betula*, and *Morus* continue to occur occasionally in trace amounts. *Celtis*, *Acer rubrum*, *A. Negundo*, and *Fagus* first appear in Zone 2. *Tilia* occurs at only two levels in the core even though it grows throughout the state (Steyermark, 1963). Because *Tilia* is insect pollinated, it usually does not occur in significant quantities.

Except for a low of 0.9% at 282 cm, *Cephalanthus* ranges from 2-7% to 152 cm, then decreases to 2.1% at 122 cm. From 122 cm to 48 cm, *Cephalanthus* values range from 3-6%, decrease slightly and then increase again to the top of Zone 2 (Plate 3). *Salix* remains relatively consistent throughout Zone 2. *Cornus*, *Rubus*, *Sambucus*, *Vitis*, and *Alnus* continue to occur in trace amounts. *Corylus* and several other shrubs first appear in this zone (Plate 3).

With the transition to Zone 2, Gramineae (Plate 3) increases sharply from 13.6 to 24%. Although they fluctuate, Gramineae values decrease to 164 cm, then remain relatively constant (between 7 and 10%) to 110 cm and increase significantly to 25% at 90 cm. Gramineae values then decline to 13% at 35 cm and increase to 23% at the top of Zone 2. Cyperaceae follows a similar trend but at much lower values (Plate 3). *Ambrosia*, Tubuliflorae, Chenopodiaceae/Amaranthaceae, and *Equisetum* occur as consistent minor components throughout Zone 2 (Plate 3). Consistently occurring in trace amounts are *Artemisia*, Cruciferae, *Impatiens*, and *Urtica*-type pollen (Plate 3). Several other herbs appear sporadically in Zone 2.

There are several problems with the pollen concentration and influx rates (see Pollen Concentration and Influx Rates), but it is interesting to note that almost all pollen concentrations and influx rates decrease significantly after 1400 years B.P., yet the taxa that are or can be associated with wetland habitats share a less significant decrease in concentration and influx rates (Plates 4, 5).

Cephalanthus, *Salix*, Gramineae, Cyperaceae, *Sparganium*, and *Potamogeton* all show this trend (Plates 4, 5). In addition to the small but significant increase in *Sparganium* and *Potamogeton* (Plate 3), this trend may indicate an increase in moisture available to Buttonbush Bog during this period. Two grasses, an indeterminable species of *Poa* and *Sphenopholis intermedia* Rydb., were found growing in or at the edge of the bog (Appendix II.A). Steyermark (1963:99) reported that two varieties of *Poa annua* L. (*P. annua* var. *aquatica* Aschers and *P. annua* var. *reptans* Haussknecht) grow submerged in springs and spring branches in Missouri. *Poa annua* var. *reptans* is known to occur in Carter County (Steyermark, 1963, Map 132).

Preliminary analysis of macrofossils from large organic residue retained from pollen extraction (Appendix III.A) of this section of the core yielded several seeds of *Eleocharis obtusa* (Willd.) Schultes, *Polygonum punctatum* Ell., and *Carex*, all wet ground plants. The presence of these taxa together with the evidence mentioned above suggests an increase in available moisture to the bog. Thus, the rise in Gramineae and Cyperaceae pollen is probably a result of increased abundance of wet ground grasses and sedges in the bog.

The *Pinus* values in Zone 2 are more consistent with the modern pollen rain values recorded by King (1973) and this study (Plate 1) than those in Zone 1. Gramineae percentages for most of Zone 2 are higher than those reported by King (1973), Peterson (1978), and this study (Plate 1), but are close to those from northwest Iowa (Van Zant, 1976, 1979). *Carya* values are consistent with those reported in this study, and those of King (1973) and Peterson (1978).

Zone 2 is interpreted as representing the Pine-Oak forest described for the area by Braun (1950) and Steyermark (1959, 1963). Although hickory is a minor component, this zone represents K  chler's Oak-Hickory-Pine forest (1964).

Zone 3: Ambrosia Pollen Assemblage Zone (0-17 cm)

A small but significant change in pollen content occurs in Zone 3 (Plate 3). The increase in *Ambrosia* from 1% in Zone 2 to 5.6% can be attributed to land clearance by pioneers and to the beginning of lumbering and modern agriculture in the vicinity of Buttonbush Bog. Sauer (1920) reported that initial settlement of the area began about 1820. The first permanent settlement was established on the Current River in Ripley County (adjacent to Carter County to the south) in 1819 when there were already permanent settlements in Oregon County to the southeast. By 1867, Carter County had become important as a producer of pine lumber (Sauer, 1920); nevertheless, a large stand of virgin pine still occurs 30 km north of Buttonbush Bog.

The pollen concentration and influx rates of *Ambrosia* and other weedy plants, such as Chenopodiaceae/Amaranthaceae and *Artemisia*, show slight increases in this zone (Plates 4, 5). The small *Ambrosia* rise found at Buttonbush Bog instead of the customary large and distinct *Ambrosia* rise usually found in conjunction with land clearance in the Midwest (King, 1981; Huber, 1980, 1985; Van Zant, 1976, 1979) may be the result of a shielding effect by the uncut forest still surviving in the area today. Another possible explanation for the small *Ambrosia* rise is that *Ambrosia* may not be prominent in this area (King, 1978).

ROUND SPRING SHELTER, ROUND SPRING SITE 23SH19

Quercus and Tubuliflorae dominate the pollen spectra throughout the Round Spring Shelter core (Plate 7). In all but the uppermost sample interval, nonarboreal pollen (herbs) occur in greater than 50% of the pollen sum, and then only decrease to 47.4%. The pollen sum decreases to its lowest concentration at the same time (Plate 8). Note that the point representing the sample is plotted at the center of the interval in Plates 7, 8, and 9 and that the gap in the diagram occurs between the overlapping levels. Because there were so few *Pinus* half grains, they were combined with *Pinus echinata*-type on all the Round Spring Shelter diagrams.

The main feature of the diagram is the high percentages of Tubuliflorae pollen, which range from 28% at the bottom to 57.3% in the 26-34 cm interval, then decline to 14.5% at the top. As Tubuliflorae declines, *Pinus* concentrations (Plate 8) increase in a like manner, indicating that the increase in pine percentages (Plate 7) is real and is not masked by the high Tubuliflorae values. *Quercus* ranges from 12 to 25% and concentration varies between 420 and 1400 grains/cm³.

There are two *Ambrosia* peaks in the pollen diagram, one in the 34-37 cm sample interval, and the second above the overlapping samples in the 7-12 cm interval. The upper rise is probably the usual rise found in conjunction with land clearance and pioneer settlement (King, 1981; Huber, 1980, 1985; Van Zant, 1976, 1979). The *Ambrosia* rise is accompanied by a decrease in *Quercus*, also indicating clearing of the forest for lumber and field cultivation. *Quercus* concentrations (Plate 9) decrease from 840 grains/cm³ in the 12-16 cm interval to 425 grains/cm³ in the 7-12 cm interval. The lower *Ambrosia* rise, however, may indicate a prolonged period of Indian occupation of Round Spring Site 23SH19. The hearth stone (Fig. 26) found at 65 cm in the core and the archaeological material retrieved from the test unit (see Specific Site Descriptions) indicate at least intermittent use of the shelter for most of the pollen record.

Based on the 95% confidence interval, there are very few significant differences in percentage values between the overlapping sample intervals (Plate 9), whereas concentrations sometimes vary considerably (Plate 8). Indeterminable degraded pollen is present in significant quantities, ranging

from 7 to 27% (Plate 7). Concentration of degraded pollen varies from 180 to 1550 grains/cfn³. The high values of degraded pollen are not unexpected since soil oxidation and soil microbes make dry soils less conducive to pollen preservation (King and others, 1975).

The pollen diagram from Round Spring Shelter is divided into two zones (Plate 7). The boundary between the zones is hard to discern because it occurs in the overlapping intervals. Therefore, for convenience, sample intervals occurring below the gap in the diagram (Plate 7) are referred to as Zone 1 and sample intervals above the gap as Zone 2.

Zone 1 is characterized by a decline in *Chenopodiaceae/Amaranthaceae*, *Cyperaceae*, *Gramineae*, *Fraxinus quadrangulata*, and *Pinus* towards the top, while at the same time *Juniperus*, *Dryopteris*, and *Lycopodium* increase (Plate 7). Zone 2 contains the second *Ambrosia* rise and is characterized by a decline in *Tubuliflorae* and an increase in *Pinus*, *Carya*, and the herbs *Chenopodiaceae/Amaranthaceae*, *Cyperaceae*, and *Gramineae* all increase towards the top of this zone. *Lycopodium* and *Dryopteris* (both vascular cryptogams) also rise, but *Juniperus* declines slightly.

The pollen spectra from the Round Spring Shelter core are hard to interpret. The author was unable to find *Tubuliflorae* values this high in any archaeological or geological pollen diagrams. Lynott (written commun., 1981) reported that relic hunters have removed several burials from inside the shelter. Therefore, the pollen spectra may represent disturbed and intrusive material. Further evidence for the periodic disturbance of the sediments in the shelter is that Round Spring flooded over its confines in 1982, cutting a channel (Fig. 35) through the gravel path located on top of the natural bridge, under which the Round Spring waters flow. Floods of this nature may also have occurred in the past and greatly influenced the pollen spectra.

It is interesting to note that *Tubuliflorae* decreases as *Ambrosia* increases during the probable time of pioneer settlement, as shown in the upper part of the diagram. This may indicate that the large *Tubuliflorae* values are related to Indian use of Round Spring Shelter. A sharp decline in *Tubuliflorae* would be expected if its presence is related to Indian use of the shelter; its lack may be a result of homogenization of the sediment by relic hunters. In addition, it is possible that the



Figure 35. Photograph of the channel cut through the gravel path located on top of the natural bridge at Round Spring during the 1982 flood. Photo courtesy of M.J. Lynott, National Park Service, 1982.

gradual decline in Tubuliflorae is related to the sampling technique in which the entire interval is represented by a homogenized subsample.

None of the modern pollen rain data for this area (this study; King, 1973; Peterson, 1978) correlates well with the pollen spectra from Round Spring Shelter. This is to be expected because the site is a shelter and is not exposed to the full effects of pollen rain. Furthermore, the site has probably been influenced by culturally specific selection of certain taxa.

Keeping in mind that several factors may have influenced the pollen deposition at Round Spring Shelter, the following interpretations have been made. The Round Spring Shelter pollen spectra represent a pine-oak forest growing in the vicinity of Round Spring Site 23SH19 (Fig. 17) with ferns growing in the cooler, moister area near the spring (Fig. 16). The consistent presence of *Ambrosia* and other weedy plants probably represents disturbance created by Indian occupation at the site. The *Ambrosia* rise in Zone 2 is attributed to pioneer settlement and land clearance. Except for a possible cultural affiliation, the large Tubuliflorae values are unexplained at this time.

GOOSENECK SITE 23CT54

Quercus is the most consistent dominant pollen type in the pollen spectra from the Gooseneck Site samples. The most interesting feature of the pollen diagram is the presence of *Zea mays* (corn), which was found in seven of the eight samples and was identified solely by grain size. Kapp (1963) uses a maximum diameter of 70 micrometers as a basis of minimum size in his taxonomic pollen key. To distinguish a possible cornfield site in Arizona, Martin and Schoenwetter (1960) used 60 micrometers as their minimum size criteria for *Zea mays*. Examination of size frequencies of pollen from Mandan yellow flour, Mandan black, and Mandan Nueta corn cultivars yielded average size frequencies of 80, 69, and 69 micrometers, respectively. Based on this information, 68 micrometers was used as a minimum diameter for identification. All of the *Zea mays* grains identified were greater than 70 micrometers and some approached 100 micrometers in diameter. Preservation of the pollen at Gooseneck Site is poor and a great many of the pollen grains are partially degraded. When

identifying *Zea mays*, only grains in which a distinct pore was observed were counted. However, some of the identifications are questionable as a result of poor preservation, and the data should be used with some caution. It should also be noted that Gooseneck Site 23CT54 was once part of a farmstead where corn was raised to feed mules used in lumbering operations in the area (Potter, 1973). It is possible that *Zea mays* pollen found below the plowzone may have migrated downward by means of bioturbation or water flow and the *Zea mays* pollen found in archaeological context may not have been originally associated with Indian occupation. In all the pollen diagrams (Plates 10, 11, 12) for Gooseneck Site, *Pinus* half grains have been combined with *Pinus echinata*-type because of their low values. The pollen spectra will be discussed by provenience and the arbitrary sample numbers (Table 10) will be used for convenience.

22N/4-5E (Samples 1 and 5)

Sample 1 overlies sample 5 and they are from the A-P horizon and subsoil, respectively. The pollen spectra from sample 5 are composed of 54% herbs. Caryophyllaceae is the dominant herb (15%), followed by Umbelliferae (8.5%), and Tubuliflorae (7.3%). *Ambrosia*-type, Chenopodiaceae/Amaranthaceae, Cyperaceae, and Gramineae make up the rest of the major herbs found in this sample (Plate 10). *Cornus* (24%) is the dominant arboreal constituent in sample 5. *Quercus* occurs at a value of 11% and *Juniperus* at 5%. Eleven grains of *Zea mays* were found in this sample, comprising 2.7% of the pollen sum.

In the overlying sample (sample 1), Caryophyllaceae and Umbelliferae pollen were not present. *Cornus* values decreased from 24% in sample 5 to 1.2% in sample 1. *Juniperus* also decreased from 5 to 0.7%. However, *Quercus* increased from 11 to 29% while *Pinus* increased from 2.2 to 6.1% (Plate 10). *Ambrosia*-type, Chenopodiaceae/Amaranthaceae, and Gramineae all increased significantly in this level. Tubuliflorae decreased from 7.3 to 0.7%. *Zea mays* increased slightly to 3.1% (Plate 10).

Most of the taxa show significant differences between the samples based on the 95% confidence intervals (Plate 11) and pollen concentrations vary greatly (Plate 12).

22-23N/22-23E (Samples 2 and 6)

Samples 2 and 6 are from the midden and submidden, respectively. The submidden (sample 6) is characterized by 75% arboreal pollen (trees and shrubs). *Quercus* is the dominant pollen type (43.6%), *Pinus* is low (0.7%), and *Salix* occurs at 7.2%. *Cornus* is the second most abundant taxa at 12.4% (Plate 10). *Ambrosia*-type is the dominant herb (6.2%); the rest of the herbs occur with values of less than 5% (Plate 10).

Arboreal pollen also dominates the pollen spectrum from sample 2 (92.3%). *Salix* increases from 7.2% to 39.1%. *Quercus* decreases to 21.4%, and *Nyssa* (a minor component in sample 6) increases from 1.2 to 20.2%. *Cornus*, *Frazinus*, and *Juniperus* also decrease (Plate 10). Gramineae and Tubuliflorae increase slightly from the submidden to the midden level (Plate 10). However, the rest of the herbs found in sample 6 decrease or do not occur in sample 2. *Zea mays* is only found in the submidden and occurs at 1.5% (Plate 10). The major pollen types in these samples also show significant differences between the submidden and midden levels (Plate 11).

22-23N/24-25E (Samples 3 and 7)

Samples 3 and 7 are also from the midden and submidden, respectively. Sample 7 is dominated by herbs (58.2%), with *Ambrosia*-type (14.5%), *Lycopodium* (9.1%), and Gramineae (15%) as the major components (Plate 10).

Chenopodiaceae/Amaranthaceae (3.2%), Cyperaceae (6.9%), and Tubuliflorae (4.9%) make up the rest of the dominant herbs. *Quercus* (20.1%), *Carya* (6.1%), *Juniperus* (4.7%), *Pinus* (2.2%), *Salix* (2.7%), and *Frazinus quadrangulata* (2.2%) are the most abundant arboreal pollen (Plate 10).

Sample 3 is dominated by arboreal pollen. *Quercus* increases from 20.1% in sample 7 to 23.6% in sample 3. *Carya* decreases slightly to 4.7% whereas *Frazinus quadrangulata*, *Juniperus*, and *Salix* all increase. *Ambrosia*-type and Gramineae both show a significant decrease from sample

7 to sample 3 (Plate 11), whereas *Chenopodiaceae*/*Amaranthaceae*, *Tubuliflorae* and *Lycopodium* increase. *Zea mays* increases from 0.7% in sample 7 to 3.7% in sample 3 (Plate 10).

31-32N/12-13E (Sample 4)

Sample 4 is from the midden level of the house feature and is dominated by herbs (60%). *Lycopodium* (16.2%), *Ambrosia*-type (15.2%), and Gramineae (10.6%) are the major herbs found in this sample (Plate 10). *Quercus* accounts for 16.2% of the arboreal pollen while *Carya* (3.7%), *Juniperus* (6.1%), *Pinus* (3.9%), and *Fraxinus quadrangulata* (3.9%) make up the rest of the major arboreal pollen types. *Zea mays* comprises 1% of the pollen spectrum in Sample 4 (Plate 10).

32-33N/12-13E (Sample 8)

From the adjoining test unit of the house feature, sample 8 is also dominated by herb pollen (57.4%). *Lycopodium* (13.3%), Gramineae (12.8%), Cyperaceae (11.6%), and *Ambrosia*-type (11.1%) make up 48.8% of the pollen spectra (Plate 10). *Quercus* (14.5%) is again the dominant arboreal pollen type. *Juniperus* (6.9%), *Fraxinus quadrangulata* (4.9%), *Populus* (4.9%), *Pinus* (3.7%), and *Carya* (2.7%) comprise the rest of the major shrubs (Plate 10). Only one grain of *Zea mays* pollen occurred in sample 8.

Pollen concentrations (Plate 12) and the 95% confidence intervals (Plate 11) show significant differences between all the samples for some taxa and not for others. Large differences in concentrations of individual taxa between associated samples also occur (Plate 12). The variation in the samples is probably caused in part by differential pollen preservation. Second, some of the pollen types may be more abundant in a particular area of the site as a result of culturally specific selection by Indian occupants. Finally, the natural pollen rain may be influenced by a large influx of local pollen. The large values of *Salix* and *Nyssa* in sample 2 appear to indicate this.

The pollen spectra from Gooseneck show lower values for *Pinus*, *Quercus*, and *Fraxinus* than King's (1973) modern pollen rain study. Peterson's (1978) data do not correlate well to the Gooseneck Site either. The modern pollen collected from Gooseneck Site in this study (Plate 1) is

approximately correlative to several of the individual taxa from the samples but does not correlate well with any one particular sample (Plate 10). Although the percentage values of the major components from Gooseneck Site (*Quercus*, *Pinus*, *Fraxinus*, and *Ambrosia*-type) do not directly correlate to all the modern pollen rain data for the area, they do show a similar trend.

Potter (1973) analyzed a portion of the ethnobotanical material recovered during the 1971 excavation of Gooseneck. *Quercus* (oak), *Carya* (hickory), *Juniperus* (cedar), and *Acer* (maple) were identified from the charcoal remains. Carbonized acorn and hickory nut shells were also found associated with the site. *Vitis* (grape), *Rhus* (sumac), *Polygonum aviculare*, and one corn cupule make up the rest of the carbonized ethnobotanical material recovered by Potter (1973) from the Gooseneck Site. In addition to the carbonized material, several recent seeds were found at the site. The uncarbonized material includes *Viola*, *Vitis*, *Ulmus*, *Eleocharis*, *Plantanus occidentalis*, *Polygonum*, and *Amaranthus* seeds. In addition to Potter's (1973) study, Voleman (1980) analyzed macrobotanical material from both the 1971 and 1979-80 excavations at Gooseneck. Based on carbonized wood, nut shells, seeds, and uncarbonized recent seeds, Voleman (1980) identified the following taxa (at least to the family level) as occurring at Gooseneck Site: *Betula*, *Carpinus*, *Carya*, *Carya* cf. *cordiformis*, *Celtis*, Chenopodiaceae, *Cornus*, Cyperaceae, *Fagus*, Juglandaceae, *Juglans*, *Juglans* cf. *cinerea*, *Pinus*, *Platanus*, Polygonaceae, *Prunus*, *Prunus persica*, *Quercus*, Rosaceae, Salicaceae (includes *Salix* and *Populus*), *Ulmus*, and *Vitis*.

Potter (1973) and Voleman (1980) together reported 34 plant taxa, at various levels of identification, from the ethnobotanical remains retrieved from Gooseneck. Of these, 29 are represented, at least at the family level, in the fossil pollen record at Gooseneck, 21 to the level of genus, and 3 to species (Table 11). However, only 17 of the 34 plant taxa are represented, at least at the family level, in the modern pollen rain (Table 11). Table 11 also shows the contemporary vegetation collected by the author, which represents or may represent the taxa found at Gooseneck, depending on the level of identification of ethnobotanical remains. If a particular taxon is not represented by

TABLE 11. COMPARISON OF PLANT TAXA FOUND AT GOOSENECK SITE 23CT54

Ethnobotanical Remains	Fossil Pollen	Modern Pollen	Contemporary Vegetation Collected
^a Acer	<i>Acer saccharum</i> -type	<i>Acer saccharum</i> -type	<i>Acer</i> cf. <i>saccharum</i>
^a Amaranthus	Chenopodiaceae/ Amaranthaceae	Chenopodiaceae/ Amaranthaceae	
^a Betula		<i>Betula</i>	
^a Carpinus	<i>Ostrya</i> / <i>Carpinus</i>		<i>Carpinus caroliniana</i> var. <i>virginiana</i>
^a Carya	<i>Carya</i>	<i>Carya</i>	<i>Carya</i> sp. <i>Carya cordiformis</i>
^a Carya cf. <i>cordiformis</i>	<i>Carya</i>	<i>Carya</i>	<i>Carya cordiformis</i>
^a Celtis	<i>Celtis</i>	<i>Celtis</i>	
^a Chenopodiaceae	Chenopodiaceae/ Amaranthaceae	Chenopodiaceae/ Amaranthaceae	
^a Cornus	<i>Cornus</i>	<i>Cornus</i>	
^a Cyperaceae	Cyperaceae	Cyperaceae	<i>Carex</i> sp. <i>Cyperus</i> sp. <i>Cyperus ovularis</i>
^a Eleocharis	Cyperaceae	Cyperaceae	
^a Fagus			
^a Gramineae	Gramineae	Gramineae	<i>Arundinaria gigantea</i> <i>Digitaria sanguinalis</i> <i>Muhlenbergia</i> sp. cf. <i>Panicum</i> <i>Setaria glauca</i> <i>Uniola latifolia</i>
^a Juglandaceae	<i>Carya</i> <i>Juglans cinerea</i> <i>Juglans nigra</i>	<i>Carya</i>	<i>Carya</i> sp. <i>Carya cordiformis</i> <i>Juglans nigra</i>
^a Juglans	<i>Juglans nigra</i>		<i>Juglans nigra</i>

TABLE 11. COMPARISON OF PLANT TAXA FOUND AT GOOSENECK SITE 23CT54 (continued)

Ethnobotanical Remains	Fossil Pollen	Modern Pollen	Contemporary Vegetation Collected
¹ Juglans cf. cinerea	Juglans cinerea		
¹ Juniperus	Juniperus	Juniperus	
¹ Pinus	Pinus echinata-type	Pinus echinata-type	Pinus echinata
¹ Platanus	Platanus	Platanus	Platanus occidentalis var. occidentalis f. occidentalis
¹ Polygonaceae	Polygonum undiff. Polygonum aviculare-type Polygonum amphibium-type Polygonum hydropiperoides-type		Polygonum virginianum var. virginianum
¹ Polygonum	Polygonum undiff. Polygonum aviculare-type Polygonum amphibium-type Polygonum hydropiperoides-type		Polygonum virginianum var. virginianum
¹ Polygonum aviculare	Polygonum aviculare-type		
¹ Prunus			
¹ Prunus persica			
¹ Quercus	Quercus	Quercus	Quercus alba f. latiloba Quercus cf. prinoides var. acuminate
¹ Rhus			
¹ Rosaceae	Potentilla Rubus		Agrimonia rostellata Rubus sp.
¹ Salicaceae (includes)			
¹ Populus	Populus		
¹ Salix	Salix		
¹ Ulmus	Ulmus		
¹ Viola			
¹ Vitis	Vitis		Vitis vulpina
¹ Zea mays	Zea mays		

¹Found by Potter, K.L., 1973.¹Found by Voleman, K.C., 1980.

the contemporary vegetation collected, that does not necessarily mean it is not still growing at the site. Collection of the contemporary vegetation by the author was not exhaustive; thus any taxon not represented could easily have been missed. The contemporary vegetation added to Table 11 gives an indication of the possible similarity of the vegetation growing at the site now with the ethnobotanical remains.

Four of the samples analyzed for pollen came from the same test units and depths that were also analyzed for macrobotanical remains by Voleman (1980). The pollen and ethnobotanical remains for these samples are compared in Table 12. One of the pollen samples (sample 3) represents two different levels of a test unit examined by Voleman (1980); thus her data have been combined for the comparison (Table 12). In three of the four samples, all of the ethnobotanical taxon identified is represented by pollen. Sample 2, however, contained no pollen representative of the ethnobotanical material found.

Potter (1973) suggests that the single cupule of corn found at Gooseneck was probably brought to the site, rather than grown there. This is further substantiated by the faunal remains that Smith (1971, 1975) has interpreted as representing an October-November and January-May occupation of the site. The presence of *Zea mays* pollen indicates the presence of a cornfield at the site, which would suggest a prolonged occupation during the summer months and is thus contrary to the interpretation based on the faunal remains. However, as mentioned above, the presence of *Zea mays* pollen in archaeological context does not necessarily indicate an aboriginal cornfield. Faegri and Iverson (1975) and Birks and Birks (1980) report that vertical mixing is a problem in mildly acid soils where earthworms are present. It is likely that the corn pollen has migrated downward through the soil via bioturbation or water movement and represents modern corn agriculture.

The pollen record at the Gooseneck Site is interpreted as representing a mixed oak-hickory forest in the uplands and *Nyssa*, *Frazinus*, *Salix*, *Vitis*, *Juglans*, and *Ulmus* growing in the bottomlands. The vegetation during aboriginal occupation of Gooseneck was similar to that found in the area today, although pine may have been less abundant.

TABLE 12. COMPARISON OF ETHNOBOTANICAL MATERIAL AND FOSSIL POLLEN FROM THE SAME PROVENIENCE AT GOOSENECK SITE 23CT54

Provenience	Depth in cm	Type of Material	Amount (grams)	Taxa	Pollen Sample	Taxa represented by Fossil Pollen	Percent of the Pollen Sum
22-23N 22-23E	20-30	nutshell (C) charcoal	0.2 0.2	Juglandaceae <i>Carya</i> sp.	Sample 2		
22-23N 24-25E	20-30	nutshell (C) charcoal	0.4 0.5 0.7	Juglandaceae <i>Juglans</i> sp. <i>Pinus</i> sp.	Sample 3 20-40 cm	<i>Carya</i> <i>Juglans cinerea</i> <i>Juglans nigra</i> <i>Pinus echinata</i> -type	4.7% 0.2% 1.0% 3.2%
22-23N 24-25E	30-40	nutshell (C) charcoal	0.2 0.3	Juglandaceae <i>Juglans</i> sp.			
22-23N 24-25E	40-50	nutshell (C)	0.2	Juglandaceae	Sample 7	<i>Carya</i>	6.1%
32-33N 12-13E	20-30	nutshell (C) unidentified charred debris	2.6 16.7	Juglandaceae	Sample 8	<i>Carya</i> <i>Juglans cinerea</i>	2.7% 0.7%

Ethnobotanical material from Voleman, K.C., 1980.

C = carbonized.

PHYTOLITH ASSEMBLAGES

CONTEMPORARY VEGETATION REFERENCE SAMPLES

The contemporary plant samples collected in the study area have not been studied in detail. However all the plant parts digested have been scanned and initial presence/absence determination of phytoliths has been completed. Based on results of the quick scan (Appendix IV), 19 plant taxa have been identified as good possibilities for subsequent examination. The plant taxa chosen are from the families Cyperaceae, Gramineae, Labiatae, Leguminosae, Pinaceae, and Violaceae. The list of plants selected includes: *Arundinaria gigantea*, *Digitaria* ? *sanguinalis*, *Elymus virginicus* var. *virginicus* f. *hirsutiglumis*, *Poa* sp., *Panicum dichotomum*, *Panicum lanuginosum* var. *implication*, *Setaria glauca*, *Sphenopholis inetermedia*, *Uniola latifolia*, all Gramineae; *Helenium autumnale* and *Helianthus divaricatus* var. *divaricatus*, both composites; the legume, *Desmodium nudiflorum* f. *nudiform*; and *Viola lanceolata* var. *lanceolata*, *Pycnanthemum tenuifolium*, and *Pinus echinata*.

BUTTONBUSH BOG

The 63 sediment samples from Buttonbush Bog have been scanned and all samples contain phytoliths. Both grass and sedge phytolith types have been observed. Diatoms and fresh water sponge spicules occur throughout the core.

ROUND SPRING SHELTER, ROUND SPRING SITE 23SH19

In the Round Spring Shelter samples, phytoliths are less abundant than in the Buttonbush Bog samples. Of 14 samples scanned, sample intervals 37-42 cm and 42-48 cm contain the most phytoliths, based on relative abundance. Grass phytoliths were observed in all the Round Spring Shelter samples.

GOOSENECK SITE 23CT54

All the phytolith samples from Gooseneck Site 23CT54 contained the five major phytolith types included in the phytolith sum (Figs. 19-23). Detailed analysis of local corn varieties has not been undertaken for the study area and it has been shown that phytolith morphology from *Zea mays* is not universally standard (Piperno, 1984; Mulholland, 1985, oral comm.). Therefore, no attempt has been made to determine the presence or absence of corn based on phytolith data. However, the phytolith assemblages found in the Gooseneck Site sediments were compared with distribution of phytoliths from plant subfamilies reported by Mulholland and Rapp (1985). Table 13 shows the results of their study.

TABLE 13. DISTRIBUTION PATTERN OF GRASS SILICA PHYTOLITHS

Phytolith Categories	Long Trapezoid	Short Trapezoid	Saddle	Dumbbell & Cross
Grass Subfamilies*				
Pooideae	X	X		X
Arundinoideae		X	X	X
Chloridoideae		X	X	X
Panicoideae				X

From Mulholland, S.C., and Rapp, G., Jr., 1985.

*Grass subfamilies follow Pohl, R.W., 1978.

All four phytolith categories presented by Mulholland and Rapp (1985) were present in the phytolith samples from Gooseneck Site 23CT54, suggesting that any or all of the four grass subfamilies could have grown there. It is highly probable that the subfamily Pooideae grew at the Gooseneck Site because it is the only subfamily known at this time to contain long trapezoids. However, subsequent examination of phytolith assemblages from grass species not yet examined may prove that long trapezoids occur in subfamilies other than Pooideae.

SUMMARY AND CONCLUSIONS

The purpose of this study was the interpretation of the late Holocene vegetational history of the southeast Missouri Ozarks based on palynological data from Buttonbush Bog; Round Spring Shelter, Round Spring Site 23SH19; and Gooseneck Site 23CT54. Correlative objectives were to undertake preliminary comparisons between modern plant phytolith assemblages and fossil phytolith assemblages and to compare pollen and phytolith data from the same sediment samples as well as provide a general overview of the paleoecology and the potential natural resources available in the study locality.

This study focuses on sites in the Current River watershed. Originating at Montauk Springs in the southcentral Ozarks, the Current River flows for 225 km to the Missouri-Arkansas border and then approximately another 50 km in Arkansas before entering the Black River. In Missouri, the Current River drains an area of 5490 km², draining a basin approximately 145 km in length and 80 km at its maximum width.

Four springs of first magnitude feed the Current River; one of these is Big Spring, the largest single-outlet spring in the United States. These four springs together with other natural springs contribute approximately 60% of the Current River's flow, giving it the highest sustained base flow in the state and providing a year-round source of water for aboriginal inhabitants. In Carter and Ripley counties, the Current River valley is considered the boundary between the southeastern and southern slopes of the Salem Plateau.

The climate of the southeast Missouri Ozarks is essentially continental. The area receives cold air from Canada, warm moist air from the Gulf of Mexico, and dry air from the west, resulting in frequent weather changes. The study locality lies near the border of the Prairie Peninsula and the Southeast and Eastern Seaboard Climatic Regions. The Prairie Peninsula Climatic Region is characterized by low winter rainfall and snowfall and occasional summer droughts. The Southeast and Eastern Seaboard Climatic Region is characterized by rainy winters with more rain and sunnier days in the summer, creating weather patterns that have great ecological impact on this area.

The southeast Missouri Ozarks are located within the stable Mid-Continent area and have as their principal structural feature the Ozark uplift or Ozark dome. This area has been the focal point of mild repeated uplifts since Precambrian time and has been a land area continuously since Pennsylvanian time.

Bedrock in the area is composed primarily of chert-bearing dolomite of Cambrian and Ordovician age. Some sandstone occurs in the Ordovician Gasconade and Roubidoux formations. Thus, chert and sandstone sources were available for aboriginal chipped and ground stone tool manufacture. Common in both the Cambrian Eminence Formation and the Ordovician Gasconade Formation are caves and springs that would have provided shelter and fresh water.

Faunal resources native to the study locality and available to early pioneers and the Indians who preceded them included deer, bison, elk or wapiti, wolves, bears, panther or mountain lion, wildcats or bobcats, beaver, otter, muskrat, mink, raccoon, opossum, skunk, fox and gray squirrels, fox, chipmunk, and cottontail rabbits. Turkey, quail, passenger pigeon, and occasional other fowl were also available.

Fish in the area included bass, jack salmon or wall-eyed pike, sunfish, stone cat, suckers, channel cat, bullhead cat, mud cat, buffalo, crappie, short-nosed gar, eel, and minnows. Bullfrogs and turtles also inhabited the area.

The Ozark region is characterized by a diversified flora composed of pine-oak, oak-hickory, or oak-hickory-pine woodlands, although hickory is a minor component. The forest flora with its herbaceous components belong to the Carolinian flora with a slight dominance of southern species. Within the study locality, five edaphic plant associations have been delineated by Steyermark (1963) based on physical, chemical, and local moisture conditions:

1. Sugar Maple-Bitternut Hickory Association (*Acer saccharum-Carya cordiformis*)
2. Sugar Maple-White Oak Association (*Acer saccharum-Quercus alba*)
3. Oak-Hickory Association (*Quercus-Carya*)
4. Oak-Pine Association (*Quercus-Pinus echinata*)
5. White Oak-Red Maple Association (*Quercus alba-Acer rubrum*)

These associations, however, may differ somewhat from the flora present at the time of pioneer settlement. In addition to the forest flora, prairie flora often inhabit forest openings and glades. Within the southeast Missouri Ozarks many microclimates and microenvironments occur, each possessing a characteristic assemblage of plants that add greatly to the number of potential resource plants found within the area. Appendix V is a list of potential native food and beverage plants of the southeast Missouri Ozarks.

In the study locality the Buttonbush Bog core contains a record of the last 3100 years of late Holocene vegetation. During this time the area has undergone little vegetational change. Zone 1 from the basal sediments of the core is dominated by oak and represents a mixed oak forest with minor components of pine and hickory. The low values of pine in this zone indicate that it is just beginning to become locally established in this area of the southeastern Ozarks. In Zone 2, pine values increase as pine becomes a more dominant component of the Ozark flora. Hickory continues to be a minor but consistent component of the Ozark forest. *Cephalanthus* was probably growing in or around the edge of the bog throughout Zone 2. Pollen influx and concentration decline and (after 1400 B.P.) drop significantly. The increase in aquatics along with the sedges and grasses indicates an increase of moisture available to Buttonbush Bog after 1400 B.P. The pollen record suggests that a pine-oak forest occurred in the area throughout Zone 2. Zone 3 is marked by a small but significant increase in *Ambrosia*-type, the result of pioneer settlement and land clearance beginning about 1820.

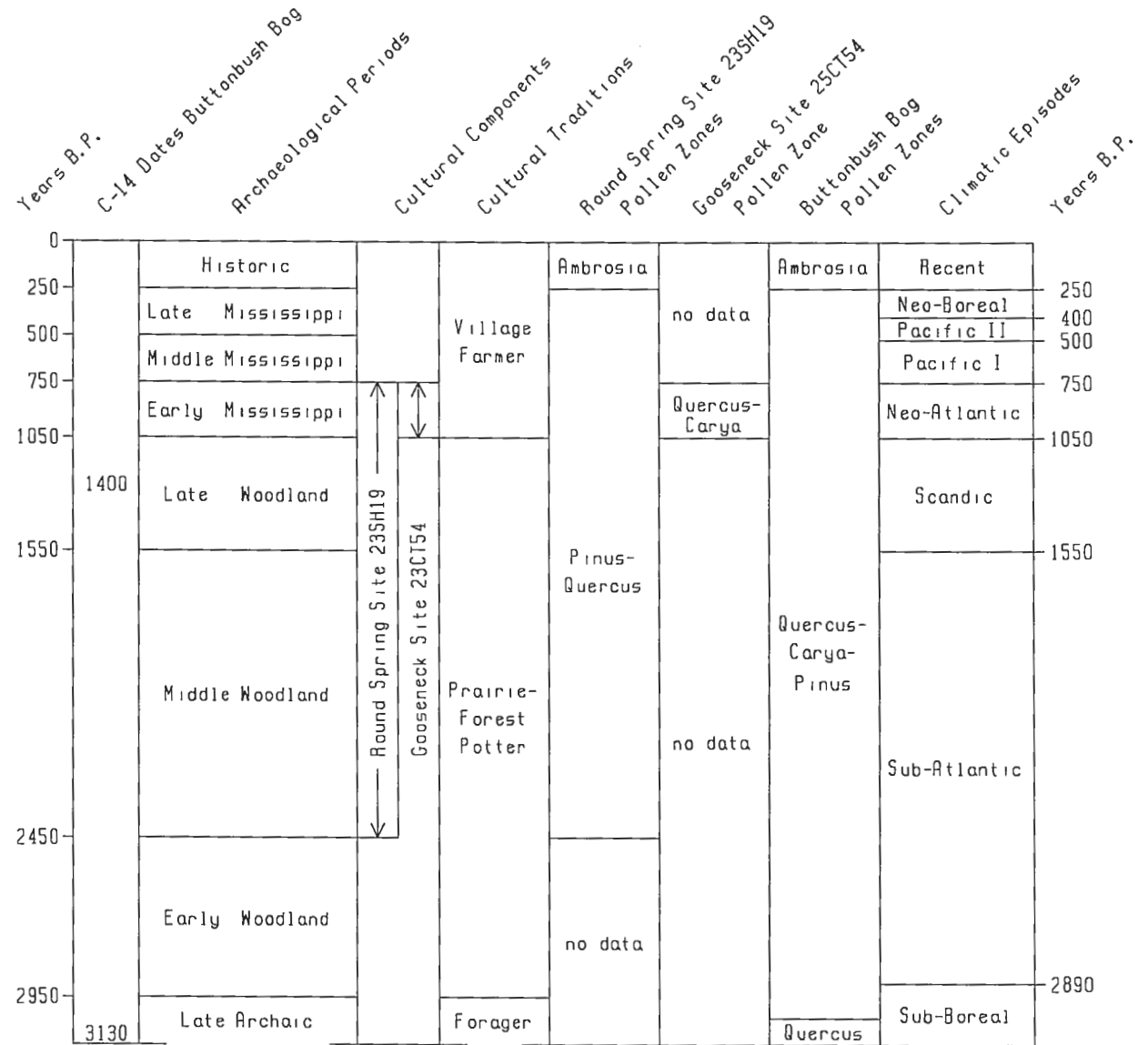
The Round Spring Shelter, Round Spring Site 23SH19 pollen spectra are also indicative of a pine-oak forest. The rise of *Ambrosia*-type pollen in the lower zone is attributed to Indian occupation of the site; pioneer settlement and land clearance is probably the cause of the *Ambrosia*-type rise in Zone 2. The large values of Tubuliflorae pollen found at Round Spring Shelter may be related to Indian occupation of the site. However, the reason for the Tubuliflorae abundance is unknown at this time.

The pollen record from the eight Gooseneck Site samples suggests a mixed oak-hickory forest growing in the uplands near the site and ash, willow, grape, elm, walnut, hickory, and tupelo growing in the river bottoms. Based on phytolith assemblages from the same samples, species of four subfamilies of grasses may have grown at the site. The pollen data from the archaeological samples correlate well with the ethnobotanical remains reported by Potter (1973) and Voleman (1980) for Gooseneck.

Zea mays pollen has been found at Gooseneck Site 23CT54; whether the pollen is associated with modern agriculture or connected with Indian occupation is uncertain. Faunal evidence from Gooseneck (Smith, 1971, 1975) suggests late fall and early spring occupation of the site. This indicates that the Gooseneck site was not occupied during the summer growing season, and the corn pollen is probably associated with modern agriculture on the site.

The vegetational records contained in the sediments of the sites studied are associated with eight archaeological time periods beginning with the late Late Archaic period and continuing to the present. Figure 36 is a summary of the environmental and cultural records found at the sites investigated in this locality of the southeast Missouri Ozarks.

Figure 36. Summary of the environmental and cultural records found at Buttonbush Bog, Round Spring Shelter, Round Spring Site 23SH19, and Gooseneck Site 23CT54.



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APPENDIX I: PLANT NAMES

Scientific and colloquial names of plants mentioned in the text and on the pollen diagrams are listed alphabetically by the scientific name. Plant names follow Steyermark for all taxa that occur in Missouri.

Acer L., Maple

Acer Negundo L., Box elder

Acer rubrum L., Red maple

Acer saccharum Marsh., Sugar maple

Agrimonia rostellata Wallr., Agrimony

Alnus serrulata (Ait.) Willd., Common alder

Amaranthaceae, Amaranth Family

Amaranthus L., Amaranth

Amaranthus tuberculatus (Moq.) Sauer, Water hemp

Ambrosia L., Ragweed

Amorpha L., False indigo

Artemisia L., Wormwood

Arundinaria gigantea (Walt.) Champ., Cane

Arundinoideae, subfamily of Gramineae

Arundo donax L., Giant reed

Athyrium Filix-femina (L.) Roth., Lady fern

Betula L., Birch

Betulaceae, Birch Family

Boraginaceae, Borage Family

Botrychium Sw., Grape fern

Brassica L., Mustard

Bromus inermis Leyss, Brome grass

Carex L., Sedge

Carpinus L., Hornbeam, Blue beech

Carpinus caroliniana Walt. var. *virginia* (Marsh.) Fern., Blue beech

Carya Nutt., Hickory

Carya cordiformis (Wang.) K. Koch, Bitternut hickory

Caryophyllaceae, Pink family

Castanea Mill., Chestnut

Celtis L., Hackberry

Cephalanthus occidentalis L. var. *occidentalis* f. *occidentalis*, Buttonbush

Cheilanthes feei Moore, Fee's lip-fern

Chenopodiaceae, Goosefoot Family

Chenopodiineae, Chenopodiaceae and Amaranthaceae Families

Chenopodium L., Goosefoot

Chloridoideae, subfamily of Gramineae

Cornus L., Dogwood

Corylaceae, Birch Family

Corylus L., Hazelnut

Cruciferae, Mustard Family

Cucurbita L., Squash

Cucurbita pepo L., Squash and pumpkin

Cyperaceae, Sedge Family

Cyperus L., Umbrella sedge

Cyperus ovularis (Michx.) Torr, Hedgehog club rush

Cystopteris bulbifera (L.) Bernh., Bladder fern

Cystopteris fragilis (L.) Bernh., Common fragile fern

Dennstaedtia punctiloba (Michx.) Moore, Hay scented fern
Desmodium nudiflorum (L.) DC f. *nudiflorum*, Tick trefoil
Digitaria sanguinalis (L.) Scop., Crabgrass
Dirca palustris L., Leatherwood
Dryopteris Adans., Shield fern
Dryopteris Goldiana (Hook.) Gray, Goldie's fern
Eleocharis R. Br., Spike rush
Eleocharis obtusa (Willd.) Schultes var. *obtusa*, Spike rush
Elymus virginicus L. var. *virginicus* f. *hirsutiglumus* (Scribn.) Fern, Wild rye
Ephedra nevadensis S. Wats., Mormon tea
Ephedra torreyana S. Wats., Mormon tea
Epilobium coloratum Biehler, Willow herb
Equisetaceae, Horsetail Family
Equisetum L., Horsetail
Eragrostis cilianensis (All.) Lutati, Stinkgrass
Ericaceae, Heath Family
Eucalyptus L'Herit., Used for pollen spike
Fagus L., Beech
Fraxinus americana L., White ash
Fraxinus pennsylvanica Marsh., Red ash
Fraxinus quadrangulata Michx., Blue ash
Galium L., Bedstraw
Gramineae, Grass Family
Helenium autumnale L., Sneezeweed
Helianthus L., Sunflower
Helianthus divaricatus L. var. *divaricatus*, Sunflower

Hippuris L., Mare's tail
Humulus L., Hops
Ilex L., Holly
Impatiens L., Touch-me-not
Iva L., Marsh elder
Iva ciliata Willd., Marsh elder
Iva xanthifolia Nutt., Marsh elder
Juglandaceae, Walnut family
Juglans cinerea L., Butternut
Juglans nigra L., Walnut
Juncaceae, Rush Family
Juniperus L., Cedar, Juniper
Labiatae, Mint Family
Larix Mill., Larch
Leguminosae, Pea Family
Lemna L., Duckweed
Liguliflorae, subfamily of Compositae
Liliaceae, Lily Family
Liquidambar L., Sweet gum
Lycopodium L., Clubmoss
Lycopus L., Bugle weed
Menispermum L., Moonseed
Menyanthes L., Buckbean
Mitella L., Miterwort
Morus L., Mulberry
Muhlenbergia Schreb., Muhly

Myrica L., Meadow fern
Myriophyllum heterophyllum Michx., Water milfoil
Nuphar Sm., Yellow pond lily
Nuphar luteum (L.) Sibth. & Sm., Yellow pond lily
Nymphaea L., Water lily
Nyssa L., Tupelo
Osmunda L., Royal fern
Ostrya Scop., Hop hornbeam, Ironwood
Panicoideae, subfamily of Gramineae
Panicum L., Panic grass
Panicum dichotomum L., Panic grass
Panicum lanuginosum Ell. var. *implicatum* (Scribn.) Fern, Panic grass
Phalaris L., Canary grass
Phleum pratense L., Timothy
Phlox L., Phlox
Phoradendron flavescens (Pursh) Nutt., Mistletoe
Picea Dietr., Spruce
Pinus L., Pine
Pinus echinata Mill, Short leaf pine
Pinus strobus L., White pine
Planera J.F. Gmel., Water elm
Plantago L., Plantain
Platanus L., Sycamore
Platanus occidentalis L. var. *occidentalis* f. *occidentalis*, Sycamore
Poa L., Blue grass
Poa annua L., Annual blue grass

Poa annua var. *aquatica* Aschers, Blue grass

Poa annua var. *replans* Haussknecht, Blue grass

Polygonaceae, Buckwheat family

Polygonum L., Knotweed

Polygonum amphibium L., Water smartweed

Polygonum aviculare L., Knotweed

Polygonum hydropiperoides Michx., Wild water pepper

Polygonum lapathifolium L., Knotweed

Polygonum punctatum Ell., Water Smartweed

Polygonum virginianum L. var. *virginianum*, Virginia knotweed

Polypodium L., Polypody

Polypodium vulgare L., Common Polypody

Polystichum Roth.

Polystichum acrostichoides (Michx.) Schott, Christmas fern

Pooideae, subfamily of Gramineae

Populus L., Poplar

Portulacaceae, Purslane Family

Potamogeton L., Pondweed

Potentilla L., Cinquefoil

Proserpinaca palustris L., Mermaid weed

Prunus L., Plum, Cherry

Prunus persica L., Peach

Pteridium Gleditch, Bracken fern

Pteridium aquilinum (L.) Kuhn, Bracken fern

Pycnanthemum tenuifolium Schrad., Slender mountain mint

Quercus L., Oak

Quercus alba L. f. *latiloba* (Sarg.) Palmer & Steyerl., White oak
Quercus prinoides Willd. var. *accuminata* (Michx.) GL., Chestnut oak
Ranunculaceae, Crowfoot Family
Ranunculus L., Buttercup
Rhamnaceae, Buckthorn Family
Rhus L., Sumac
Robinia L., Locust
Rosaceae, Rose Family
Rubus L., Raspberry, Blackberry
Rumex L., Dock
Sagittaria L., Arrowhead
Salicaceae, Willow family
Salix L., Willow
Sambucus L., Elderberry
Saxifragaceae, Saxifrage Family
Selaginella Beauv., Spikemoss
Setaria glauca (L.) Beauv., Yellow foxtail
Setaria lutescens (Weigel.) Hubb., Yellow foxtail
Sium L., Water parsnip
Smilacina Desf., False Solomon's seal
Sparganium L., Bur-reed
Sphagnum L., Sphagnum moss
Sphenopholis intermedia Rydb., Wedge grass
Stachys L., Hedge nettle
Taxodium Rich., Bald cypress
Taxus L., Yew

Thalictrum L., Meadow rue
Thelypteris palustris Schott, Marsh fern
Tilia L., Basswood
Tilia americana L., Basswood, Linden
Tillandsia usneoides L., Spanish moss
Tsuga (Endl.) Carr., Hemlock
Tubuliflorae, subfamily of Compositae
Typha latifolia L., Common cattail
Ulmus L., Elm
Umbelliferae, Parsley Family
Uniola latifolia Michx., Broadleaf uniola
Urtica L., Nettle
Utricularia L., Bladderwort
Viola L., Violet
Viola lanceolata L. *lanceolata*, Lance-leaved violet
Vitis L., Grape
Vitis vulpina L., Winter grape
Woodsia obtusa (Spreng.) Torr., Blunt-lobed Woodsia
Xanthium L., Cocklebur
Zea mays L., corn

APPENDIX II.A: CONTEMPORARY VEGETATION COLLECTED AT BUT-
TONBUSH BOG

Compositae

Silphium Asteriscus L., Starry rosin-weed

Solidago ulmifolia Muhl. var. *ulmifolia*, Elm-leaved Goldenrod

Cornaceae

Cornus florida L. f. *florida*, Flowering dogwood

Cyperaceae

Carex lupulina Muhl., Hop sedge

Fagaceae

Quercus alba L. f. *latiloba* (Sarg.) Palmer & Steyererm., White oak

Gramineae

Panicum dichotomum L. Panic grass

Panicus lanuginosus Ell. var. *implicatum* (Scribn.) Fern., Panic grass

Poa sp., Blue grass

Sphenopholis intermedia Rydb., Wedge grass

Hypericaceae

Hypericum mutilum var. *parriflorum* (Willd.) Fern., Dwarf St. John's-wort

Hypericum punctatum L. var. *punctatum*, St. John's-wort

Juncaceae

Juncus tenuis Willd. f. *tenuis*, Path rush

Lauraceae

Sassafras albidum (Nutt.) Nees var. *molle* (Raf.) Fern., Red sassafras

Leguminosae

Desmodium nudiflorum (L.) DC. f. *nudiflorum*, Tick trefoil

Nymphaeaceae

Nuphar luteum (L.) Sibth. & Sm., Yellow pond lily

Pinaceae

Pinus echinata Mill., Short-leaf pine

Polygonaceae

Polygonum sp., Knotweed

Polygonum hydropiperoides Michx., Wild water pepper

Rubiaceae

Cephalanthus occidentalis L. var. *occidentalis* f. *occidentalis*, Buttonbush

Violaceae

Viola lanceolata L. var. *lanceolata*, Lance-leaved violet

Vitaceae

Vitis sp., Grape

APPENDIX II.B: CONTEMPORARY VEGETATION COLLECTED AT OZARK
SINK POND

Anacardiaceae

Rhus copallina L. var. *latifolia* Engler, Dwarf sumac

Bignoniaceae

Campsis radicans (L.) Seem., Trumpet creeper

Compositae

Ambrosia artemisiifolia L. var. *elatior* (L.) Descourt f. *elatior*, Ragweed

Helenium autumnale L., Sneezeweed

Helianthus divaricatus L. var. *divaricatus*, Sunflower

Solidago sp., Goldenrod

Vernonia Baldwini Torr. var. *interior* (Small) Schubert, Ironweed

Cornaceae

Cornus racemosa Lam., Gray dogwood

Nyssa cf. *sylvatica* Marsh., Black gum

Cyperaceae

Carex sp., Sedge

Carex squarrosa L., Sedge

Eleocharis obtusa (Willd.) Schultes var. *obtusa*, Spike rush

Ericaceae

Vaccinium sp., Blueberry

Fagaceae

Quercus sp., Oak

Quercus alba L. f. *latiloba* (Sarg.) Palmer & Steyererm., White oak

Gramineae

Panicum dichotomum L., Panic grass

Juglandaceae

Carya ovalis (Wang.) Sarg., Red hickory

Labiatae

Monarda russeliana Nutt., Horsemint

Pycnanthemum tenuifolium Schreb., Slender mountain mint

Lauraceae

Sassafras albidum (Nutt.) Nees var. *molle* (Raf.) Fern., Red sassafras

Leguminosae

Apios americana Medic., Groundnut

cf. *Baptisia* sp., False indigo

Cassia fasciculata Michx. var. *fasciculata* f. *fasciculata*, Partridge pea

Desmodium sp., Beggar's ticks

Desmodium nudiflorum (L.) DC. f. *nudiflorum*, Tick trefoil

Melastomataceae

Rhexia virginica L. var. *virginica*, Meadow Beauty

Rubiaceae

Cephalanthus occidentalis L. var. *occidentalis* f. *occidentalis*, Buttonbush

Ulmaceae

Ulmus cf. *alta* Michx., Winged elm

APPENDIX ILC: CONTEMPORARY VEGETATION COLLECTED AT ROUND
SPRING SITE 23SH19

Aceraceae

Acer nigrum Michx. f. *nigrum*, Black maple

Alismaceae

Sagittaria sp., Arrowhead

Anacardiaceae

Rhus glabra L. var. *glabra*, Smooth sumac

Campanulaceae

Campanula americana L., Tall bellflower

Compositae

Cichorium Intybus L., Chickory

Solidago ulmifolia Muhl. var. *ulmifolia*, Elm-leaved Goldenrod

Convolvulaceae

Ipomoea pandurata (L.) G.F.W. Mey. f. *leviuscula* Fern., Morning glory

Cornaceae

Cornus obliqua Raf., Swamp dogwood

Cruciferae

Nasturtium officinale R. Br., Watercress

Cupressaceae

Juniperus virginiana L., Red cedar

Cyperaceae

Cyperus refractus Engelm., Sedge

Fagaceae

Quercus alba L., White oak

Gramineae

Elymus virginicus L. var. *virginicus* f. *hirsutiglumis* (Scribn.) Fern., Wild rye

Haloragidaceae

Myriophyllum heterophyllum Michx., Water milfoil

Lauraceae

Lindera Benzoin (L.) Blume, Spice bush

Leguminosae

Cercis canadensis L. var. *canadensis*, Eastern redbud

Trifolium pratense L., Red clover

Najadaceae

Potamogeton nodosus Poir., Pondweed

Plantaginaceae

Plantago lanceolata L., English plantain

Rosaceae

Fragaria virginiana Duchesne var. *illinoensis* (Prince) Gray,

Wild strawberry

Rutaceae

Ptelea trifoliata L. var. *trifoliata* f. *trifoliata*, Hoptree

Saxifragaceae

Hydrangea arborescens L. var. *Deamii* St. John f. *Deamii*, Wild hydrangea

Penthorum sedoides L., Dutch stonecrop

Sparganiaceae

Sparganium americanum Nutt., Bur-reed

Umbelliferae

Daucus carota L. f. *carota*, Queen Ann's lace

Violaceae

Viola sp., Violet

APPENDIX II.D: CONTEMPORARY VEGETATION COLLECTED AT GOOSE-
NECK SITE 23CT54

Aceraceae

Acer cf. *saccarum* Marsh, Sugar maple

Betulaceae

Carpinus caroliniana Walt var. *virginiana* (Marsh) Fern., Blue beech

Campanulaceae

Campanula americana L., Tall Bellflower

Caprifoliaceae

Symphoricarpos occidentalis Hook., Wolfberry

Compositae

Ambrosia artemisiifolia L., Common ragweed

Bidens connata Muhl. var. *petiolata* (Nutt.) Farw., Beggars ticks

Eupatorium purpureum L., Joe-Pye weed

Cyperaceae

Carex sp., Sedge

Cyperus sp., Umbrella sedge

Cyperus ovularis (Michx.) Torr., Sedge

Ebenaceae

Diospyros virginiana L. var. *platycarpa* Sarg. f. *atra* Sarg., Persimmon

Fagaceae

Quercus alba L. f. *latiloba* (Sarg.) Palmer & Steyermark, White oak

Quercus cf. *prinoides* Willd. var. *acuminata* (Michx.) GL., Chestnut oak

Gramineae

Arundinaria gigantea (Walt.) Chapm., Cane

Digitaria sanguinalis (L.) Scop., Crabgrass

Muhlenbergia sp., Muhly

cf. *Panicum* sp., Panic grass

Setaria glauca (L.) Beauv., Yellow foxtail

Uniola latifolia Michx., Spike grass

Juglandaceae

Carya sp., Hickory

Carya cordiformis (Wang.) K. Koch, Pignut Hickory

Juglans nigra L., Black walnut

Labiatae

Unidentified species

Blephilia hirsuta (Pursh) Benth. var. *hirsuta*, Wood mint

Prunella vulgaris L., Self-heal

Lauraceae

Sassafras albidum (Nutt.) Nees var. *albidum*, White sassafrass

Leguminosae

Amorpha fruticosa L., False indigo

Cassia fasciculata Michx. var. *robusta* (Pollard) Macbr., Partridge pea

Desmodium sp., Beggar's ticks

Lespedeza cuneata (Dumont) G. Don, Sericea Lespedeza

Trifolium repens L. f. *repens*, White Dutch clover

Liliaceae

Smilax rotundifolia L., Greenbrier

Menispermaceae

Menispermum canadense L., Moonseed

Nymphaeaceae

Nuphar luteum (L.) Sibth. & Sm. subsp. *macrophyllum* (Small) Beal, Yellow pond lily

Orchidaceae

Unidentified species

Oxalidaceae

Oxalis stricta L., Yellow wood sorrel

Pinaceae

Pinus echinata Mill., Short-leaf pine

Plantaginaceae

Plantago Rugelii Dcne. var. *Rugelii*, Plantain

Plantanaceae

Plantanus occidentalis L. var. *occidentalis* f. *occidentalis*, Sycamore

Polygonaceae

Polygonum virginianum L. var. *virginianum*, Virginia knotweed

Ranunculaceae

Anemone virginiana L., Thimbleweed

Rosaceae

Agrimonia rostellata Wallr., Agrimony

Rubus sp., Blackberry

Saururaceae

Saururus cernus L., Lizard's-tail

Scrophulariaceae

Gerardia grandiflora Benth., Gerardia

Tiliaceae

Tilia americana L., Basswood, Linden

Vitaceae

Parthenocissus quinquefolia (L.) Planch. f. *quinquefolia*, Virginia creeper

Vitis vulpina L., Winter grape

APPENDIX II.E: CONTEMPORARY VEGETATION COLLECTED AT INCIDENTAL SITES

Cypress Swamp, Butler County

Hamamelidaceae

Liquidamber styraciflua L., Sweet gum

Taxodiaceae

Taxodium distichum (L.) Rich. var. *distichum* f. *distichum*, Bald cypress

Lewis Lake, Shannon County

Magnoliaceae

Liriodendron tulipifera L., Tulip tree

Malvaceae

Hibiscus lasiocarpus Cav., Rose mallow

Phytolaccaceae

Phytolacca americana L. Pokeweed

Van Buren, Carter County

Leguminosae

Gleditsia triacanthos L. var. *triacanthos*, Honey locust

APPENDIX IIIA: LABORATORY PROCEDURES: POLLEN PREPARATION PROCEDURE FOR SMALL QUATERNARY SEDIMENT SAMPLES

1. Before starting, make sure you know laboratory safety procedures. Wear laboratory apron during entire process. Wear gloves and/or face shield for specified steps. If you wish to do a pollen concentration or pollen influx study, follow steps 2 and 3. If not, go to step 4.
2. Obtain a preweighed *Eucalyptus* pollen tablet for each sample you run. Record the weight of the tablet on a POLLEN CONCENTRATION DATA SHEET.
3. Put the selected tablet into a graduated 15-ml centrifuge tube properly labeled for its respective interval. NOTE: Be sure the selected tablet is in the proper test tube and all pollen concentration information is recorded on the POLLEN CONCENTRATION DATA SHEET. Add just enough 10% HCl (hydrochloric acid) to dissolve the CaCO_3 cement in the tablet (usually less than 3 ml).
4. Place 1-2 (5 in exceptional cases) ml of sediment in the centrifuge tube. Add 10 ml distilled water. Stir until sediment is well dispersed. Centrifuge 5 minutes at setting of 5. Measure carefully and record the volume of the sediment on the data sheet if pollen concentration is wanted. Decant.
5. Add 10 ml of phytolith/pollen dispersant (NaPO_3). Stir, centrifuge 5 minutes at setting of 5, and decant. (The liquid may be cloudy.) If sample is rich in clay-sized mineral particles, repeat until liquid is fairly clear.
6. Add 10 ml of 10% KOH, stir, and place in hot water bath for 6 to 10 minutes. Centrifuge and decant. Add distilled water, stir and wash with distilled water through precleaned Coors filtering crucible into small beaker. Swirl water and sediment in beaker and pour off water and fine particles into centrifuge tube, leaving sand behind. The nalgene (plastic) tube will be needed in step 9. Stir sample thoroughly to begin to remove humic acids as in step 7. Centrifuge and decant. Repeat until swirled water is clear. Rinse the residue from the Coors filtering crucible and beaker into a glass jar, label, allow to evaporate at room temperature, and cap. With this step, macrofossils and seeds from the sample are obtained and stored.
7. Rinse sample in centrifuge tube with distilled water, stir, centrifuge, and decant. Repeat until water is fairly clear (i.e., no longer "humic" brown in color). This will take several repetitions.
8. If sediment is calcareous, add 2 ml of 10% HCl and observe reaction. If little bubbling occurs, add 3 ml more HCl. If bubbling is violent, wait until it stops, centrifuge, decant, and again add 2 ml 10% HCl. Repeat until little bubbling occurs. Add 10% HCl up to 5-ml level, place in hot bath. Again watch bubbling; if bubbling subsides or 10 minutes have passed, centrifuge, decant. Rinse with distilled water, stir, centrifuge, decant.

STEPS 9 THROUGH 14 SHOULD BE DONE UNDER THE FUME HOOD.

9. **USE RUBBER GLOVES AND FACE PROTECTOR FOR THIS STEP:** The samples should now be in nalgene (plastic) test tubes. Add 5 ml of concentrated (48%) HF (HYDROFLUORIC ACID) slowly to each of the 15-ml tubes. (A few extra ml of HF may be used if a large quantity of sand is present.) Heat in boiling water bath for 15 minutes. Fill each tube to 10 ml with distilled water, stir (using wood stirring rods only), centrifuge, and decant supernatant into the large nalgene bottle marked "Used HF." Repeat if necessary. If a white precipitate remains, fluorite crystals have grown. To remove fluorite crystals, add 5 to 10 ml 10% HCl, place in hot water bath (at setting 5 on the small hot plate) for 5 minutes, centrifuge, and decant; repeat hot HCl treatment until precipitate disappears.

10. Rinse with distilled water, stir, centrifuge, decant.
11. Transfer sediment with a little distilled water back into 15-ml graduated glass centrifuge tubes. Centrifuge and decant.
12. Rinse with 10 ml concentrated GLACIAL ACETIC ACID (HAc); stir, centrifuge, decant, repeat, repeat one more time.
13. **USE RUBBER GLOVES AND FACE PROTECTOR FOR THIS STEP:** Carefully add 5 to 8 ml of acetolysis solution to each sample. Acetolysis solution should be prepared in one batch just prior to use; it is composed of 1 part concentrated SULFURIC ACID (H_2SO_4) added to 9 parts ACETIC ANHYDRIDE. Thus for a 6-tube batch, **slowly** add 3 ml of concentrated H_2SO_4 to 27 ml of acetic anhydride, using a small graduated cylinder. There should be little or no acetolysis solution left over from a run; it cannot be stored. (Any left over should be poured into a large volume of cold tap water and this slowly poured down the drain.) Stir the sediment (with a wood or glass stirring rod) in the acetolysis solution and heat in the water bath (setting 5 on small hot plate) for two minutes. Then fill each tube within 1 cm of its lip with cold glacial acetic acid; stir thoroughly, centrifuge, and decant.
14. Add 3 ml of 10% KOH + 7 ml distilled water to each tube, stir thoroughly, centrifuge, decant.
15. Rinse with distilled water; stir, centrifuge, decant.
16. Add 5 ml of phytolith/pollen dispersant; stir. The suspension is then passed through a fresh 10-micrometer Nitex screen (mounted on a frame placed on a receptacle connected to a vibrator) to separate the clay fraction (which is washed into the filter flask) from the pollen-bearing fraction (which is caught by the Nitex screen). Rinse filtrate with distilled water until waste fluid is nearly clear. Rinse the material on the screen into a labeled 250-ml beaker. Pour into glass 15-ml centrifuge tube, centrifuge, and decant until all the sample is transferred. Rinse once with distilled water, centrifuge, and decant.
17. Add 10 ml of 95% ETHANOL (ETOH) and 10 drops of safranin stain (1% aqueous solution) to each tube, stir thoroughly. Let stand 5 minutes, centrifuge, and decant.
18. Add 10 ml of absolute ETOH to each tube; stir, centrifuge, decant.
19. Add just enough TERTIARY BUTYL ALCOHOL (TBA) to each tube to pour the sample. Stir thoroughly. (The TBA usually must be warmed slightly to melt it; do this in a warm water bath or by running warm tap water over the closed container.)
20. Transfer the pollen sample in TBA to a standard 1-dram labeled glass vial. Flush tube with a fine jet of TBA to aid in transferring pollen sample to glass vial. Centrifuging will require adding extra inserts (an empty 1-dram vial) to the centrifuge sleeves to support lips of the vials above the sleeves. The supernatant TBA is carefully withdrawn from the vial with an eye dropper and discarded. The eye dropper must be cleaned between each sample with 95% ETOH. This is done by filling the eye dropper twice with ETOH.
21. When all the sample is transferred to the vial and all the TBA removed, add enough silicone oil to cover pollen residue (about twice as much oil as there is pollen), and allow the vial to stand open overnight in a warm, dust-free place to let the TBA evaporate. (This step should not be prolonged as the sample might dry out. A few hours may suffice.)
22. Make sure **both** the vial and the cap are labeled, using indelible ink pen.

Revised 5 October 1982 by J.K. Huber and D.L. Balach

Revised 6 February 1984 by J.K. Huber

Archaeometry Laboratory, Duluth, Minnesota

APPENDIX III.B: LABORATORY PROCEDURES: POLLEN PREPARATIONS FROM LARGE-VOLUME SEDIMENT SAMPLES

1. Before starting, make sure you know laboratory safety procedures. Wear laboratory apron during entire process. Wear gloves and/or face shield for specified steps. If you wish to do a pollen concentration or pollen influx study, follow steps 2 and 3. If not, go to step 4.
2. Obtain a preweighted *Eucalyptus* pollen tablet for each sample you run. Record the weight of the tablet on a POLLEN CONCENTRATION DATA SHEET and in the Pollen Analysis Logbook.
3. Put the selected tablet into a graduated 50-ml centrifuge tube properly labeled for its respective interval. NOTE: Be sure the selected tablet is in the proper test tube and all pollen concentration information is recorded on the POLLEN CONCENTRATION DATA SHEET. Add just enough 10% HCl (hydrochloric acid) to dissolve the CaCO_3 cement in the tablet (usually less than 3 ml). After the *Eucalyptus* tablet has dissolved, add approximately 1 ml of acetone to neutralize the HCl.
4. Weigh out 30 ± 0.5 g of sediment on the beam balance. Place the sediment in the centrifuge tube. Add distilled water as needed. Stir until sediment is well dispersed. If the sediment reacts when placed in the tube, add acetone or other base. Centrifuge 5 minutes at setting of 5. Measure carefully and record the volume of the sediment on the data sheet, if pollen concentration is wanted. Decant.
5. Transfer the sample to a labeled fleaker beaker and half fill with distilled H_2O if necessary. Shake the fleaker beaker by hand until all the sediment is well dispersed. Sieve the sample through the 20-mesh (0.833 mm) and the 80-mesh (0.177 mm) screens into a plexiglass cylinder. Transfer the contents of the plexiglass cylinder into the appropriately labeled fleaker beaker. Rinse the residue from both screens into a glass jar, label, allow to evaporate at room temperature, and cap. With this step, macrofossils and seeds from the interval are obtained and stored.
6. Add a few drops of 50% HCl-50% ETOH to the suspension. If there is no reaction, proceed to step 7. If there is a significant evolution of CO_2 gas, then the sample will have to be acidified. To accomplish this, slowly add 50% HCl-50% ETOH drop by drop until no more bubbling occurs.
7. Add 20 ml of phytolith/pollen dispersant to the fleaker beaker. Shake well.
8. The disaggregated suspension is then passed through a new Nitex screen (mounted in the frame made from a large wash bottle and positioned on the shaker in the plexiglass tube). This is to separate the clay fraction (which is washed into the plexiglass cylinder) from the pollen-bearing silt fraction (which remains on the screen). For best results fill the screwtop portion of the wash bottle with sample. Then add distilled H_2O to the frame until the tapered area is filled. Stir with a medium size glass stirring rod while the shaker is operating. Add distilled H_2O to the sample until the water is clear as it comes out of the screen. Transfer the silt fraction from the Nitex screen to a 250-ml beaker using distilled H_2O . Repeat this procedure until the clay has been removed from the entire sample.
9. Transfer the sample to a labeled nongraduated, tapered 50-ml centrifuge tube. Centrifuge, decant, and repeat until all the sample is in the tube. Fill the tube to approximately 30 ml with absolute ETOH, stir, centrifuge, decant. Repeat. This should remove all traces of water.

STEPS 10 THROUGH 12 SHOULD BE DONE UNDER THE FUME HOOD.

USE RUBBER GLOVES AND GAS MASK FOR STEPS 10 THROUGH 12.

10. Split the silt sample between two 50-ml pyrex centrifuge tubes. Add 25 ml of heavy liquid to each tube. (The liquid is a mixture of tetrabromethane and absolute ETOH made up to a specific gravity of 2.0 by using the heavy liquid hydrometer). The sediment and heavy liquid are thoroughly mixed and then centrifuged for 10 minutes at setting 4.
11. Each 50-ml tube should now exhibit a sedimented fraction composed of almost all silt-sized mineral grains and a suspended fraction composed of all organic material, including pollen. The **entire** heavy liquid volume in both tubes is decanted into a glass-stoppered 250-ml Erlenmeyer flask. The volume in the flask is brought to 150 ml using absolute ETOH, which dilutes the sp.g. of the heavy liquid below 1.5. Shake the sample well and transfer to one (clean) 50-ml pyrex tapered centrifuge tube. Centrifuge these tubes again to sediment out the organic fraction. Decant the diluted heavy liquid into the large nalgene bottle marked "Used, Unfiltered Heavy Liquid" for recycling. Discard the mineral fraction that remains in the original centrifuge tubes.
12. Using absolute ETOH, wash the organic fraction from the 50-ml tubes into a single 15-ml **Nalgene** centrifuge tube. Wash the sample twice more in absolute ETOH to remove all traces of heavy liquid.
NOTE: NEVER DUMP ANY QUANTITY OF THE HEAVY LIQUID MIXTURE DOWN THE DRAIN. Before putting glassware that contained heavy liquid into the washpan, rinse it once with ethanol.
13. Add 10 ml of 10% KOH, stir, and place in hot water bath for 6 to 10 minutes. Centrifuge and decant.
14. Rinse sample in centrifuge tube with distilled water, stir, centrifuge, and decant. Repeat until water is fairly clear (i.e., no longer "humic" brown in color).

STEPS 15 THROUGH 21 SHOULD BE DONE UNDER THE FUME HOOD.

15. **USE RUBBER GLOVES AND FACE PROTECTOR FOR THIS STEP:** Add 5 ml of concentrated (48%) HF (HYDROFLUORIC ACID) slowly to each of the 15-ml nalgene centrifuge tubes. Heat in boiling water bath for 15 minutes. Fill each tube to 10 ml with distilled water, stir (using wood stirring rods only), centrifuge, and decant supernatant into the large nalgene bottle marked "Used HF." If a white precipitate remains, fluorite crystals have grown. To remove fluorite crystals, add 5 to 10 ml 10% HCl, place in hot water bath (at setting 5 on the small hot plate) for 5 minutes, centrifuge, and decant; repeat hot HCl treatment until precipitate disappears.
16. Rinse with distilled water, stir, centrifuge, decant.
17. Transfer sediment with a little distilled water back into 15-ml graduated glass centrifuge tubes. Centrifuge and decant.
18. Rinse with 10 ml concentrated GLACIAL ACETIC ACID (HAc); stir, centrifuge, decant, repeat one more time.
19. **USE RUBBER GLOVES AND FACE PROTECTOR FOR THIS STEP:** Carefully add 5 ml acetolysis solution to each sample. Acetolysis solution should be prepared in one batch just prior to use; it is composed of 1 part concentrated SULFURIC ACID (H_2SO_4) added to 9 parts ACETIC ANHYDRIDE. Thus for a 6-tube batch, slowly add 3 ml of concentrated H_2SO_4 to 27 ml of acetic anhydride, using a small graduated cylinder. There should be little or no acetolysis solution left over from a run, since it cannot be stored. (Any leftover should be poured into a large volume of cold tap water and slowly poured down the drain.) Stir the sediment (with a wood or glass stirring rod) in the acetolysis solution and heat in

the water bath (setting 5 on small hot plate) for two minutes. Then fill each tube within 1 cm of its lip with cold glacial acetic acid; stir thoroughly, centrifuge, and decant.

20. Add 3 ml of 10% KOH + 7 ml distilled water to each tube, stir thoroughly, centrifuge, decant.
21. Rinse with distilled water, stir, centrifuge, decant.
22. Add 5 ml of 95% ETHANOL (ETOH) and 10 drops of safranin stain (1% aqueous solution) to each tube, stir thoroughly. Let stand 5 minutes, centrifuge, and decant.
23. Add 10 ml of absolute ETOH to each tube; stir, centrifuge, and decant.
24. Add just enough TERTIARY BUTYL ALCOHOL (TBA) to each tube to pour the sample. Stir thoroughly. (The TBA usually must be warmed slightly to melt it; do this in a warm water bath or by running warm tap water over the closed container.)
25. Transfer the pollen sample in TBA to a standard 1-dram labeled glass vial. Flush tube with a fine jet of TBA to aid in transferring the pollen sample to the glass vial. Centrifuging will require adding extra inserts (an empty 1-dram vial) to the centrifuging sleeves to support the lips of the vials above the sleeves. The supernatant TBA is carefully withdrawn from the vial with an eye dropper and discarded. The eye dropper must be cleaned between each sample, by filling the eye dropper twice with 95% ETOH.
26. When all the sample is transferred to the vial and all the TBA removed, add enough silicone oil to cover the pollen residue (about twice as much oil as there is pollen), and allow the vial to stand open overnight in a warm, dust-free place to let the TBA evaporate. (This step should not be prolonged as the sample might dry out. A few hours may suffice.)
27. Make sure **both** the vial and the cap are labeled, using an indelible ink pen.

Written 5 October 1982 by J.K. Huber
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APPENDIX III.C: LABORATORY PROCEDURES: POLLEN PREPARATION FROM MOSS POLSTERS FOR MODERN POLLEN RAIN STUDIES

1. Before starting, make sure you know laboratory safety procedures. Wear laboratory apron during entire process. Wear gloves and/or face shield for specified steps.
2. If possible, place entire moss polster into a labeled 2-liter plastic bottle.
3. Add equal parts 95% Ethanol (ETOH) and distilled water until plastic bottle is full.
4. Agitate on the roller agitator for 15 minutes.
5. Flush the moss polster with distilled water through a filter funnel into a labeled beaker. This step is designed to remove the pollen from the moss polster.
6. The moss polster and any other material in the filter funnel should then be placed in a second labeled beaker to dry. When dry, the moss polster is saved for reference.
7. Flush the pollen residue sample collected in step 5 through a Coors filtering crucible into a labeled beaker. This step is designed to remove the smaller pieces of moss from the pollen that passed through the filter funnel.
8. The moss and any other material in the Coors filtering crucible should then be placed in the beaker containing the moss polster saved in step 6.
9. Swirl water and pollen residue in beaker and pour off water and fine particles into a 15-ml glass centrifuge tube, leaving sand size particles behind. Centrifuge 5 minutes at setting 5, and decant. Repeat until all the pollen sample is transferred to the tube.
10. Rinse samples with distilled water, centrifuge, and decant.
11. Add 10 ml of 10% KOH, stir with glass or wood stirring rods and place in a hot water bath at setting 4 for 6 to 10 minutes. Centrifuge and decant. Discard the supernatant.
12. Rinse sample in centrifuge tube with distilled water, stir, centrifuge, and decant. Repeat until water is fairly clear (i.e., no longer "humic" brown in color). This will require several repetitions.
13. If sediment is calcareous, add 2 ml of 10% HCl and observe reaction. If little bubbling occurs, add 3 ml more HCl. If bubbling is violent, wait until it stops, centrifuge, decant, and again add 2 ml 10% HCl. Repeat until little bubbling occurs. Add 10% HCl up to 5-ml level, place in hot bath. Again watch bubbling; if bubbling threatens to overflow tube, add a few drops of acetone or other base. When bubbling subsides or 10 minutes have passed, centrifuge, decant. Rinse with distilled water, stir, centrifuge, decant.
14. With a small amount of distilled H₂O, transfer the samples to 15-ml graduated nalgene (plastic) centrifuge tubes, centrifuge, and decant.

STEPS 15 THROUGH 20 SHOULD BE DONE UNDER THE FUME HOOD.

15. **USE RUBBER GLOVES AND FACE PROTECTOR FOR THIS STEP:** The samples should now be in nalgene (plastic) centrifuge tubes. Slowly add 5 ml concentrated (48%) HF (HYDROFLUORIC ACID) to each of the 15 ml. (A few extra ml of HF may be used if a large quantity of sand is present.) Heat in boiling water bath for 15 minutes. Fill each tube to 10 ml with distilled water, stir (using wood stirring rods only), centrifuge, and decant supernatant into the large nalgene bottle marked "Used HF." Repeat if necessary. If a white

precipitate remains, fluorite crystals have grown. To remove fluorite crystals, add 5 to 10 ml 10% HCl, place in hot water bath (at setting 5 on the small hot plate) for 5 minutes, centrifuge, and decant; repeat hot HCl treatment until precipitate disappears.

16. Rinse with distilled water, stir, centrifuge, decant.
17. Transfer sediment with a little distilled water back into 15-ml graduated glass centrifuge tubes. Centrifuge and decant.
18. Wash sample with 10 ml of Concentrated GLACIAL ACETIC ACID (HAc), stir, centrifuge, and decant. (Save supernatant to dilute the used supernatant in step 19 before discarding.) REPEAT.
19. **USE RUBBER GLOVES AND FACE PROTECTOR FOR THIS STEP:** Carefully add 5 ml of acetolysis solution to each sample. A couple of ml extra may be used for larger samples with abundant organic material. Acetolysis solution should be prepared in one batch just prior to use; it is composed of 1 part concentrated SULFURIC ACID (H_2SO_4) added to 9 parts ACETIC ANHYDRIDE. Thus for a 6-tube batch, slowly add 3 ml of concentrated H_2SO_4 to 27 ml of acetic anhydride, using a small graduated cylinder. There should be little or no acetolysis solution left over from a run, as it cannot be stored. (Any extra should be poured into the supernatant from step 12.) Stir the sediment with a glass or wood stirring rod and heat in the water bath (setting 5 on a small hot plate) for 2 to 5 minutes, depending on how much organic material is present. Fill each tube within 1 cm of its lip with cold glacial acetic acid; stir thoroughly, centrifuge, and decant into the supernatant from step 18. Discard this supernatant by pouring it into the drain along with plenty of water.
20. Add 3 ml of 10% KOH + 7 ml distilled water to each tube, stir thoroughly, centrifuge, decant.
21. Rinse with distilled water; stir, centrifuge, decant.
22. Add 5 ml of phytolith/pollen dispersant; stir. Then pass the suspension through a fresh 10-micrometer Nitex screen (mounted on a frame placed on a filtering flask connected to an aspirator) to separate the clay fraction (which is washed into the filter flask) from the pollen-bearing fraction (which is caught by the Nitex screen). Rinse filtrate with distilled water until waste fluid is nearly clear. Rinse the material on the screen into a labeled 250-ml beaker. Pour into glass 15-ml centrifuge tube, centrifuge, and decant until all the sample is transferred. Rinse once with distilled water, centrifuge, and decant.
23. Add 10 ml of 95% ETHANOL (ETOH) and 10 drops of safranin stain (1% aqueous solution) to each tube, stir thoroughly. Let stand 5 minutes, centrifuge, and decant.
24. Add 10 ml of absolute ETOH to each tube; stir, centrifuge, decant.
25. Add just enough TERTIARY BUTYL ALCOHOL (TBA) to each tube to pour the sample. Stir thoroughly. (The TBA usually must be warmed slightly to melt it; do this in a warm water bath or by running warm tap water over the closed container.)
26. Transfer the pollen sample in TBA to a standard 1-dram labeled glass vial. Flush tube with a fine jet of TBA to aid in transferring pollen sample to glass vial. Centrifuging will require adding extra inserts (an empty 1-dram vial) to the centrifuge sleeves to support lips of the vials above the sleeves. The supernatant TBA is carefully withdrawn from the vial with an eye dropper and discarded. The eye dropper must be cleaned between each sample with 95% ETOH. This is done by filling the eye dropper twice with the ETOH.

27. When all the sample is transferred to the vial and all the TBA removed, add enough silicone oil to cover pollen residue (about twice as much oil as there is pollen), and allow the vial to stand open overnight in a warm, dust-free place to let the TBA evaporate. (This step should not be prolonged as the sample might dry out. A few hours may suffice.)
28. Make sure **both** the vial and the cap are labeled, using indelible ink pen.

Written 6 October 1982 by J.K. Huber
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**APPENDIX III.D: LABORATORY PROCEDURES: LABORATORY TECHNIQUE
FOR DETERMINATION OF ORGANIC CARBON AND CAR-
BONATE BY LOSS-ON-IGNITION**

1. Initial Drying

- (a) Place sample in 100-ml beaker, set in oven, and dry at 60°C for about 8 hours (i.e., overnight). Sample weight is variable, and depends primarily on how much material is available. Usually a sample should have a net weight of 2-5 g.

2. Sample Preparation: Obtaining the Dry Sample Weight

- (a) Use a mortar and pestle to powder the sample.
- (b) Transfer material to a preweighed ceramic crucible.
- (c) Dry sample in the oven for **one** hour at 90-100°C.
- (d) Remove sample from oven and place in desiccator. Allow sample to cool.
- (e) Weigh the oven-dried sample in the crucible.
 - i. Use the analytical balance and weigh to five decimal places (00.00000).
 - ii. This provides the "crucible and dry sample weight." Record this value on the TOTAL ORGANIC CARBON AND CARBONATE DATA SHEET.

3. Subjecting the Oven-dried Sample to Two Burn Events

- (a) **Burn 1:** The first burn allows calculation of the total organic carbon upon ignition at 550°C.
 - i. Place dried sample and crucible in the furnace.
 - ii. Heat sample at 550°C for **one** hour (see step 5).
 - iii. Place sample in desiccator, and allow to cool to room temperature.
 - A. Sample should stand overnight in desiccator to reach room temperature (or for at least 3-4 hours).
 - iv. Weigh sample and crucible on analytical balance.
 - A. (a) The value is the "crucible and residue after Burn 1" weight. Subtracting this value from the "crucible and dry sample weight" provides the "Total Organic Carbon Lost" for the sample.
- (b) **Burn 2:** The second burn allows the calculation of total CO₂ lost on ignition at 1,000°C, which is used to estimate the weight % carbonate in the sample.
 - i. Place sample and crucible in the furnace.
 - ii. Heat at 1,000°C for **one** hour (see step 5).
 - iii. Place sample and crucible in dessicator, allow sample to cool to room temperature.
 - A. Sample should remain in the desiccator until it reaches room temperature (i.e., overnight).
 - iv. Weigh the sample and crucible on the analytical balance.
 - A. The value obtained is the weight of the "crucible + residue after Burn 2." Record this value on the TOTAL ORGANIC CARBON AND CARBONATE DATA SHEET.

- B. The "Total CO₂ Lost" is a measurement of the CO₂ evolved from carbonate minerals present in the sample. It is the weight loss between 550 and 1,000°C and is calculated by subtracting the weight of the "crucible + residue after Burn 1."

4. Calculation of Weight Percents

- (a) Weight % Organic Carbon = $\frac{\text{Total Organic Carbon Lost}}{\text{Dry Sample Weight}} \times 100$
(b) Weight % CO₂ Lost = $\frac{\text{Total CO}_2 \text{ Lost}}{\text{Dry Sample Weight}} \times 100$
(c) Weight % Carbonate = $\frac{\text{Weight of \% CO}_2 \text{ Lost}}{\text{Fraction of CO}_2 \text{ in CaCO}_3 (0.44)}$

5. How to Use Manual Control on Furnace for Organic Weight Percents

- (a) at 550°C
- To heat up: usually set control at about 5 (unless you need to heat it quickly; then set it on high).
 - To maintain: the average temperature is around 2. At the start of the "run," set at 2.2 or slightly more if necessary. If the oven is kept heated for long periods of time, set around 1.7.
- (b) at 1000°C
- To heat up: set on high.
 - To maintain: the average temperature is just below 4.7. At the beginning set around 5 and as it heats for longer periods, keep it around 4.5.
- (c) other hints
- Check at 15-minute intervals.
 - If you need to be gone for long periods of time, keep furnace at a slightly lower temperature than the average.
 - GENERAL RULE: As time of use increases, decrease the setting of the control knob.

Written July 1982 by J.K. Huber and C.L. Hill
Revised October 1982 by J.K. Huber and D.J. Selness
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APPENDIX III.E: LABORATORY PROCEDURES: MICROSTRATIGRAPHIC TECHNIQUE OF GRAIN SIZE ANALYSIS: A METHOD TO OBTAIN PERCENTAGES OF SAND, SILT, AND CLAY

NOTE: Make sure all tare weights and sample information are recorded on the SEDIMENT GRAIN SIZE ANALYSIS DATA SHEET before starting the procedure.

I. Sample Selection

1. Obtain a dry weight sample of approximately 7.5 g and weigh on the Mettler balance to 4 decimal places. Record the weight on the SEDIMENT GRAIN SIZE ANALYSIS DATA SHEET. To obtain 7.5 g, the sample may have to be split and/or dried in a calcium chloride desiccator, or in shallow pans in a drying oven at a low temperature (do not heat at temperatures that will bake the clays or alter the mineralogical character of the muds). If the sample is to be analyzed only for the mineralogical size fractions and not for all constituents composing the sample, treatment in hydrogen peroxide might be used to remove the organic materials. This will, however, likely change the mineralogical composition of the fine fraction if X-ray diffraction samples are to be taken for the sediment. Shell and pottery fragments in some samples may also have to be removed if only the mineralogical components of the sample are to be analyzed.
2. Place the 7.5-g subsample into a 200-ml nalgene bottle, fill 2/3 full with distilled water, and label.

II. Sieving (Coarse Fraction Analysis)

3. Wet sieve to separate the coarse fraction (gravel and sand) from the fine fraction (silt and clay) using the 750-ml vacuum cylinder.

Start the suction in the 750-ml vacuum cylinder. Then place the sieve on top. There should be a rubber "O" ring between the bottom sieve and the rim of the vacuum cylinder. Now shake the bottle to get the sediment into suspension. Holding the sieve firmly on the cylinder with one hand, making a tight seal, pour the sample in a little at a time, and rinse the sample bottle thoroughly, making sure all the sample goes into the sieve. When all the sample has been sieved, turn off the vacuum and rinse any sample adhering to the bottom of the screen into the 750-ml vacuum cylinder. Remove the rubber "O" ring, put the sieve in a petri dish, and place in the oven to dry overnight (16 hours). The fine fraction can now be transferred from the vacuum cylinder to the 1000-ml settling cylinder. Rinse all particles into the settling cylinder. The label from the sample bottle should be transferred to the 1000-ml settling cylinder.

4. The next day, the sieve can be removed from the oven and allowed to cool for at least one half hour. The cooling allows the heat-expanded screen to resume its true size. Place the 4.0 phi screen into a stainless steel pan of like diameter. Dust any material remaining in the petri dish back into the sieve. On top of the sieve place a stainless steel cover of the same diameter as the sieve.

The stack should now be strapped onto the electric shaker and shaken for one half hour. After this, remove the stack and rap it vigorously on the counter for about three minutes. This serves to loosen any fine fraction grains still clinging to either the larger grains or the sieve itself. Once loose, these particles fall and are caught in the bottom pan. From there they should be dusted into the 1000-ml settling cylinder with the rest of the silt and clay.

Weigh the sieve on the Mettler analytic balance, and enter the weights on the data sheet. (Prior to this, the weight of the sieves when clean and dry should also have been entered in the space provided on the data sheet).

III. Pipetting (Fine Fraction Analysis)

The second half of the analysis is determination of the percentages of silt and clay. This is accomplished by withdrawing two 20-ml samples of the fine fraction at timed intervals as they settle out of solution.

5. Using the mouth pipette, add **exactly** 20 ml of $\text{Na}(\text{PO}_3)_6$ dispersant (100 g/1000 ml distilled H_2O) to the sample in the 1000-ml settling cylinder. After adding dispersant to the sample, if the water level is not up to the 1000-ml mark, add distilled water until there is exactly 1000 ml in the settling cylinder.
6. There is a set of special beakers for this procedure in a maroon box labeled "Microstratigraphic Technique Beakers." The box contains twelve sets of two beakers each. Each set has an etched label, and the sets are alphabetically labeled from "AS" to "LS" (the "S" stands for "short method," another name commonly used around the laboratory for this particular method of analysis). In each set there is a 4.0 ϕ and an 8.0 ϕ beaker. The original weight of the dry, clean beakers should be entered on the data sheet.
7. The settling cylinder containing the sample and dispersant should be placed in the constant temperature water bath (set at 22°C), and allowed to come to equilibrium. This usually takes about three to four hours. Now the withdrawals can be made. Place a set of beakers and a large container filled with distilled water next to the water bath, and set the calculator/stop watch to zero. Shake the settling cylinder to suspend all of the sediment for 1 minute. When all the material is suspended, right the cylinder and put it back in the constant temperature water bath, and **at the same time** start the stop watch. Remove the stopper on the cylinder and insert the mouth pipette exactly 20 ml below the surface of the sample water (the space between the uppermost green tape marks on the shaft of the pipette). **Do not release the spring clamp on the pipette yet.** When the stop watch reads 20 seconds, open the clamp and withdraw exactly 20 ml of sample. Close off the clamp, withdraw the pipette from the cylinder, and empty the contents into the 4.0 ϕ sample beaker (this is the silt/clay fraction). From the container of distilled water (a large nalgene beaker works well), withdraw at least 20 ml of water with the same pipette and empty it into the 4.0 ϕ sample beaker also. This is done to rinse any remaining clay particles out of the pipette. Put the beaker in the oven to dry overnight.
8. Repeat the same operation for the 8.0 ϕ sample at 1 hour 51 minutes exactly, at a depth of **10 cm** below the surface of the sample liquid (the middle pair of green tape marks on the shaft). When finished, place the sample in the oven to dry.
9. If X-ray diffraction analysis is to be done, re-suspend the sample after taking the 8.0 ϕ sample, and take another 8.0 ϕ sample 1 hour and 51 minutes later. Put the X-ray diffraction sample in a 30-ml nalgene bottle, and transfer the label from the settling cylinder to the sample bottle.
10. The next day remove the beakers from the oven, cool them in the open air, and weigh on the Mettler balance to 4 decimal places. Enter the weights on the data sheet.

IV. Data Calculation

11. This part of the procedure is included on the SEDIMENT GRAIN SIZE ANALYSIS DATA SHEET (see following pages).

Written February 1983 by C.L. Hill
Archaeometry Laboratory, Duluth, Minnesota

SEDIMENT GRAIN SIZE ANALYSIS DATA SHEET

(Microstratigraphic Technique)

Sample I.D.:_____

Date analysis begun:_____ Analyst:_____

Dry weight of sample:_____

Sieve code: 1 2 3 4 5 6

Sieve weighing date:_____

Sample weight plus tare weight:_____ Sand fraction

tare weight:_____ Raw data

Sample weight:_____

Beaker set: AS BS CS DS ES FS

Phi size: 4.0 phi (taken at 20 cm, 20 sec.)

Beaker weighing date:_____

Sample weight, plus tare weight, plus dispersant weight:_____

tare weight:_____

dispersant #:_____ dispersant weight:_____

Sample weight:_____

Phi size: 8.0 phi (taken at 10 cm, 1 hr 51 min)

Beaker weighing date:_____

Sample weight, plus tare weight, plus dispersant weight:_____

tare weight:_____

dispersant #:_____ dispersant weight:_____

Sample weight:_____

Calculations:

Sand fraction:

Sieved sample weight = _____ X 100 % = _____ % Sand

Original dry sample weight = _____

100 % -- _____ % Sand = _____ % silt/clay (actual)

Silt/clay fraction (4.0 phi):

(Sample weight) _____ x 50 = _____ (calculated sample weight)

Calculated samp. wt. = _____ X 100 % = _____ % Silt/clay (empirical)

Original dry sample weight = _____

Clay fraction (8.0 phi):

(Sample weight) _____ x 50 = _____ (calculated sample weight)

Calculated sample weight = _____ X 100 % = _____ % clay (empirical)

Original dry sample weight = _____

Silt/clay % (empirical) -- clay % (empirical) = silt % (empirical)

_____ % -- _____ % = _____ %

Corrected values for true percentages: $a/x = b/c$

a = Silt/clay % (actual)

b = Silt/clay % (empirical)

c = Clay % (empirical)

x = Clay % (actual)

$a/x = b/c$ _____ / _____ x = _____ / _____

$bx = ac$ _____ x = (_____) (_____) _____

$x = ac/b$ x = _____ % clay (actual)

_____ % silt/clay (actual) -- _____ % clay (actual) = _____ % silt (actual)

APPENDIX III.F: EXTRACTION OF OPAL PHYTOLITHS FROM PEAT SAMPLES I

PREPARATION: Use wooden stirring rods unless otherwise noted.

1. Place approximately 2 ml of peat in a nonporous Coors crucible; add approximately 7 ml of distilled water and stir.
2. Place in oven. Heat at 350°C for one week.
3. After one week, remove samples and rehydrate with distilled water. Transfer sample with distilled water to a 15-ml centrifuge tube.
4. **IN FUME HOOD:** Add 10 ml of 10% KOH, stir and place in hot water bath with glass stirring rods for 10 minutes. Stir after removal. Centrifuge for 10 minutes at a medium setting (on a clinical centrifuge) and decant.
5. Rinse sample in centrifuge tube with distilled water, stir, centrifuge for 10 minutes, decant. Repeat until water is fairly clear (i.e., no longer "humic" brown in color).
6. Rinse with 10 ml concentrated Glacial Acetic Acid, stir, centrifuge for 10 minutes, decant. Repeat once.
7. **USE RUBBER GLOVES, APRON, AND FACE PROTECTOR FOR THIS STEP.**
IN FUME HOOD: Acetolysis solution should be prepared in one batch just prior to use; it is composed of 1 part concentrated SULFURIC ACID (H_2SO_4) added to 9 parts ACETIC ANHYDRIDE. Thus for a 6-tube batch, slowly add 3 ml of concentrated H_2SO_4 to 27 ml of acetic anhydride, using a graduated cylinder. There should be little or no acetolysis solution left over from a run since it cannot be stored. Any leftover solution should be poured into a large volume of cold tap water and slowly poured down the drain. Add 5 ml of acetolysis solution to each sample. Stir the sediment with glass stirring rod and heat in water bath for 5 minutes. Then fill each tube to within 1 cm of the lip with cold glacial acetic acid: stir thoroughly, centrifuge for 10 minutes, and decant.
8. Add 3 ml of 10% KOH and 7 ml distilled water to each tube, stir thoroughly, centrifuge for 10 minutes, and decant.
9. **IN FUME HOOD:** Add 10 ml of 10% KOH, stir, and place in hot water bath with glass stirring rods for 10 minutes. Stir, centrifuge for 10 minutes, decant.
10. Rinse sample in centrifuge tube with distilled water, stir, centrifuge for 10 minutes, decant. Repeat until water is fairly clear (i.e., no longer "humic" brown in color).
11. Add 10 ml 95% ETOH, stir, centrifuge for 10 minutes, decant. Repeat once.
12. Transfer the phytolith sample to a 1-dram glass vial using 95% ETOH. Label the vial, cover with parafilm and cap, and store.

Written June 1983 by J.K. Huber and D.J. Selness
Archaeometry Laboratory, Duluth, Minnesota

APPENDIX III.G: EXTRACTION OF OPAL PHYTOLITHS FROM SEDIMENT SAMPLES V

1. Split each sample until a subsample weighing between 2 and 3 grams is obtained. Wash and dry splitter between runs and between samples. Save one subsample for replication and use the other for the procedure. Place sample in a large-volume test tube (25 x 150 mm) and add about 30 ml 0.005N dispersant solution (sodium hexametaphosphate). Leave overnight to assure disaggregation of samples. Transfer silt and clay fractions to 250-ml beakers (400-ml if needed) by vigorously shaking each test tube, allowing settling for 20 seconds, and pouring the supernatant into the beaker. If a sample contains a larger percentage of clay it may be placed (while still in the test tube) in the ultrasonic water bath for no more than 5 minutes, in order to ensure that all the sample is completely disaggregated. Repeat the decanting procedure into the beaker until only clear distilled water is visible after 20 seconds of settling. This procedure separates the sand fraction from the silt-clay fraction containing phytoliths.
2. Bring the volume of the clay-silt suspension up to about 150 ml with distilled water. Check pH with the pH meter. If carbonates are present, add 10% HCl to dissolve carbonates and disaggregate any clay lumps. Add enough to complete disaggregation of carbonates; Mediterranean samples tend to contain many carbonates. Bring pH back to 7.0 ± 0.2 with drops of 3N NaOH and the pH meter. Add 5 ml 0.005N dispersant solution to each beaker, stir the suspension well, and after one hour carefully pour off the supernatant clay suspension. Refill to 150 ml with distilled water, add 5 ml dispersant, and repeat until the supernatant liquid is essentially clear after the one-hour settling period. At this point, silt-sized particles (including phytoliths) are the only sediment size left in the bottom of the beaker.
3. Transfer each silt fraction to a 15-ml glass centrifuge tube and centrifuge for 15 minutes at a moderate setting (4). Decant water and add 10 ml absolute ethanol. Mix thoroughly with disposable wooden stirring rods, centrifuge, and decant. Repeat to ensure that all traces of water are removed.
4. **DO STEPS 4-6 IN A FUME HOOD:** Tetrabromomethane must be handled in a fume hood. Rubber gloves and an apron are necessary precautions; a breathing apparatus is also available. Prepare a heavy liquid mixture of tetrabromomethane and absolute ethanol to a specific gravity of 2.3. Add 5 ml to each sample, adding small amounts of heavy liquid drop by drop to each of the centrifuge tubes to bring the liquid level even in all tubes; OTHERWISE, CENTRIFUGING WILL NOT BE POSSIBLE. Mix thoroughly with disposable wooden rods, and centrifuge.
5. Centrifuge all samples for 15 minutes at moderate speed (4). All mineral grains with a specific gravity greater than 2.3 will spin to the bottom; phytoliths (plus other silica particles) will remain suspended in the heavy liquid mixture.
6. Decant the supernatant heavy liquid **carefully** into a clean, dry 15-ml test tube, leaving the sedimented silt-size minerals behind. Add 10 ml absolute ethanol and mix thoroughly with disposable wooden rods. Specific gravity should be 1.5; centrifuge and decant the supernatant. Phytoliths should be in the sediment.

To ensure complete phytolith recovery, repeat separation of heavy fraction (steps 4 and 5). Decant resulting supernatant heavy liquid into previously obtained phytolith sample. Add 10 ml absolute ethanol, mix, centrifuge and decant light fraction (combine with previous light fraction).

Transfer phytolith fraction to a 15-ml tube and add 5 ml of absolute ethanol. Centrifuge and decant. Repeat to ensure removal of all heavy liquid.

If desired, heavy and light fractions can be stored and examined as a check on procedure. Wash heavy fraction twice with 100% ethanol and save washes for recovery of tetrabromomethane. Otherwise, store the light fraction in the plastic bottle marked "Used, Unfiltered Heavy Liquid" for later recovery. Mix heavy fraction well with absolute ethanol and discard.

7. Rinse 1-dram glass vials with 95% ethanol. Transfer phytolith sample using 100% ethanol and cover with a square of parafilm. Label side and top with pertinent data and cap.

Written June 1982 by J.K. Huber and D.J. Selness
Archaeometry Laboratory, Duluth, Minnesota

APPENDIX III.H: EXTRACTION OF OPAL PHYTOLITHS FROM PLANT SAMPLES VIII

1. Weigh out 0.25 g of each plant part and place in a labeled 50-ml beaker. If less than 0.25 g is used, note the weight. If possible, include a standard sample as a check on procedure. Record plant and part numbers and part description in logbook.
2. Add sufficient concentrated Alconox solution (1 g Alconox dispersed in 50 ml distilled water) to immerse each sample (usually 25 ml per sample). Allow material to soak a minimum of 10 hours. Never leave plants in Alconox for more than a day as mold may begin to form. Using tweezers, place plant parts in a 500-micrometer Coors filtering crucible on top of a widemouth Ehrlenmeyer flask. Flush material thoroughly with distilled water and return to rinsed beaker. Add sufficient dilute (10%) HCl to immerse each sample. Soak overnight. If necessary, plants may remain in HCl for up to one week. Again using the Coors crucible, flush the plant material well. Place the plant parts in labeled 15-ml centrifuge tubes. Include a blank tube in the set at this time for use as a second check on the procedure. If possible, allow samples to dry before proceeding in order to facilitate digestion. Drying methods include: low-temperature (100°C) oven, hot water bath, and air evaporation. Air evaporation is safe but time-consuming. Oven drying is quick but chars plant material.

STEPS 3 AND 4 MUST BE DONE IN AN EXHAUST HOOD USING PLASTIC APRON AND GLOVES.

3. Prepare a Schulze solution by adding 3 parts HNO_3 to 1 part saturated KClO_3 or NaClO_3 . Allow 15 to 20 ml Schulze solution per tube for the first day's digestion and 10 to 12 ml for the second day. **SCHULZE SOLUTION MUST BE MADE FRESH FOR EACH DAY OF PLANT DIGESTION.**
4. Add 10-12 ml Schulze solution to each tube using a manostat. Place a glass stirring rod in each centrifuge tube and stir to remove any air pockets that may develop. Place tubes in preheated hot water bath, leaving an empty slot for adding water. The bath should be approximately 95°C, near but not quite boiling. Distilled water must be added to the bath as needed, usually every half hour. **SINCE LOW WATER LEVELS MAY CAUSE BUMPING AND PROBABLE SAMPLE CONTAMINATION, MAINTAIN WATER LEVEL AS HIGH AS POSSIBLE THROUGHOUT DIGESTION.** The plant samples must be monitored very closely for the first hour in the bath, since convection currents often cause undigested material to rise to the top of the tube and occasionally overflow. As digestion continues and plant cells break down, less constant attention is necessary. At this point, the acid level in the tubes should be maintained at the 15-ml level. To accelerate digestion, plant parts may be broken up using the glass rod. Continue digestion, stirring frequently, until all plant material has been digested. If some plant parts digest slowly (require more than 10 hours), remove the centrifuge tubes from the bath, raise rods, and rinse with acid into the tube to remove any phytoliths adhering to the rod. Then centrifuge the samples for 15 minutes at a setting of 5 (International clinical centrifuge). If material continues floating, return the tube to the water bath without decanting. If not, decant, refill with fresh Schulze solution, and return tube to bath. Continue digestion until complete. Most samples require at least one centrifuging cycle to facilitate complete digestion.
5. **WEAR GLOVES FOR THE FIRST WASH IN THIS STEP. DO NOT MIX ETHANOL WASTE WITH SCHULZE SOLUTION AS A VIOLENT REACTION WILL OCCUR.** Add enough distilled water to tubes to equalize the liquid level and stir well. Rinse stirring rods

with distilled water while removing from centrifuge tubes. Centrifuge for 15 minutes. Decant acid and refill with distilled water; stir sample using a glass stirring rod, then centrifuge. Repeat, using wooden stirring rods. Use wooden rods for remaining washes. Decant, add 8 drops 1M KOH, and fill with distilled water. Centrifuge and decant. Rinse with distilled water, centrifuge, and decant. Repeat. Rinse 1-dram storage vials with distilled water and label. Using repeated fine spray of 95% ethanol, transfer material from tube into vial. If any solid material remains in tube, a wooden rod may be used to facilitate transfer. Rinse tube (and rod, if used) twice or until vial is full. If vial becomes full before rinsing is complete, insert an empty vial below, using a disposable pipette. Complete transfer. Label vial, cover with parafilm to prevent evaporation, and cap.

Revised 1983 by C. Rattel
Archaeometry Laboratory, Duluth, Minnesota

APPENDIX IV: PLANTS FROM THE STUDY LOCALITY THAT HAVE BEEN ANALYZED FOR PHYTOLITHS

The plants are listed alphabetically by family, genus, and species. A list of the plant parts analyzed for each plant is included with each plant entry. The PHY number is the Archaeometry Laboratory's (University of Minnesota, Duluth) phytolith reference identification number. Each plant part will have a "Y" "N" or "?" following the entry. If a plant part is followed by a "Y" then phytoliths or silica bodies have been found in that plant part. A plant part followed by an "N" indicates no phytoliths were observed on that particular slide. A "?" following an entry indicates that the phytoliths were possible contaminants (this occurs frequently in roots if soil is not completely removed before extraction) or the slide contained too much organic material, obscuring the phytoliths on the the slide if any occurred. Note that the presence or absence of phytoliths is based on a quick scan of each slide and the results may change with further investigations, especially those with no or questionable phytolith occurrences.

Aceraceae

Acer nigrum Michx, f. *nigrum*

PHY 10798

Leaf Y; Stem ?; Seed ?; Seed wings Y; Seed w/wings Y

PHY 10847

Leaf Y; Small stem Y; Large stem N

Acer cf. *saccharum* Marsh.

PHY 10604

Leaf w/petiole Y; Small branch N; Large branch ?

Alismaceae

Sagittaria sp.

PHY 10811

Leaf N; Stem N; Root N

Anacardiaceae

Rhus copallina L. var. *latifolia* Engler

PHY 10773

Leaflet N; Petiole w/leafy material N; Small stem N; Large stem N; Inflorescence N

Rhus glabra L. var. *glabra*

PHY 10803

Leaflets N; Petiole N; Large stem N; Red berries N

PHY 10805

Leaflet N; Petiole N; Immature leaflets and petiole N; Large branch N

Betulaceae

Carpinus caroliniana Walt. var. *virginiana* (Marsh.) Fern.

PHY 10600

Leaf w/petiole N; Bark and woody stem ?; Small branch ?

Bignoniaceae

Campsis radicans (L.) Seem.

PHY 10795

Leaflets N; Petiole N; Small stem-1st. year N; Small stem-2nd. year N; Large stem N;
Root N

Campanulaceae

Campanula americana L.

PHY 10608

Leaf Y; Stem Y; Inflorescence Y; Root Y

PHY 10781

Leaf ?; Small stem N; Large stem N; Inflorescence ?; Root N

Caprifoliaceae

PHY 10646

Leaf Y; Stem Y; Large stem Y; Inflorescence and seed Y

Compositae

Ambrosia artemisiifolia L.

PHY 10647

Leaf Y; Stem Y; Root Y

Ambrosia artemisiifolia L. var. *elator* (L.) Descourt f. *elator*

PHY 10796

Leaf N; Stem N; Root N

Cichorium Intybus L.

PHY 10806

Leaf N; Stem N; Inflorescence N; Root N

Eupatorium purpureum L.

PHY 10818

Leaf Y; Stem N; Inflorescence N; Root N

Helenium autumnale L.

PHY 10786

Leaf Y; Stem N; Inflorescence N; Root ?

Helianthus divaricatus L. var. *divaricatus*

PHY 10822

Leaf Y; Stem Y; Inflorescence Y; Root Y

Silphium Asteriscus L.

PHY 10843

Leaf N; Stem Y; Inflorescence Y; Root Y

Solidago sp.

PHY 10823

Leaf ?; Stem Y; Inflorescence ?; Root Y

Solidago ulmifolia Muhl. var. *ulmifolia*

PHY 10636

Leaf N; Stem N; Inflorescence N; Root N

PHY 10777

Leaf N; Stem N; Inflorescence stem N; Inflorescence N; Root N

Vernonia Baldwini Torr. var. *interior* (Small) Schubert

PHY 10641

Leaf Y; Stem Y; Inflorescence Y; Root ?

Convolvulaceae

Ipomoea pandurata (L.) G.F.W. Mey. f. *leviuscula* Fern.

PHY 10780

Leaf ?; Large stem N; Small stem N; Inflorescence N; Root N

Cornaceae

Cornus florida L. f. *florida*

PHY 10632

Leaf ?; Small stem Y; Large Stem Y; Inflorescence Y; Seed ?

PHY 10839

Leaf N; Small stem N; Large stem N; Seed N

Cornus obliqua Raf.

PHY 10834

Leaf Y; Small stem N; Large Stem Y; Inflorescence Y;

Cornus racemosa Lam.

PHY 10642

Leaf ?; Small stem Y; Stem Y; Bark Y

Nyssa cf. *sylvatica* Marsh.

PHY 10793

Leaf Y; Small branch ?; Large branch ?; Seed ?

Cruciferae

Nasturtium officinale R. Br.

PHY 10799

Leaf N; Stem N; Root N

Cupressaceae

Juniperus virginiana L.

PHY 10802

Leaf N; Small stem N; Large stem N; Berry N

Cyperaceae

Carex sp.

PHY 10619

Leaf base and sheath-brown Y; Leaf blade-green Y; Root ?

PHY 10828

Leaf Y; Root ?

Carex lupulina Muhl.

PHY 10630

Leaf Y; Stem Y; Seed ?; Root ?

PHY 10789

Leaf Y; Stem Y; Inflorescence Y; Root ?

Carex squarrosa L.

PHY 10797

Leaf Y; Stem Y; Inflorescence Y; Root Y

Cyperus sp.

PHY 10598

Leaf Y; Sheath ?; Stem ?; Inflorescence Y; Root ?

Cyperus ovularis (Michx.) Torr.

PHY 10602

Leaf Y; Sheath Y; Stem N; Inflorescence Y; Root N

Cyperus refractus Engelm.

PHY 10807

Leaf Y; Stem Y; Inflorescence Y; Root Y

Eleocharis obtusa (Willd.) Schultes var. *obtusa*

PHY 10819

Stem Y; Inflorescence Y; Root Y

Ebenaceae

Diospyros virginiana L. var. *platycarpa* Sarg. f. *atra* Sarg.

PHY 10611

Leaf N; Small branch Y; Large branch Y; Seed Y; Sepals Y

Ericaceae

? *Vaccinium* sp.

PHY 10825

Leaf Y; Stem Y; Root ?

Fagaceae

Quercus sp.

PHY 10774

Small leaf N; Large leaf N; Small branch N; Large branch N

Quercus alba L.

PHY 10800

Leaf Y; Small branch N; Large branch N

Quercus alba L. f. *latiloba* (Sarg.) Palmer & Steyermark

PHY 10601

Leaf w/petiole ?; Bark and woody stem ?; Small branch ?

PHY 10771

Leaf N; Small Stem N; Large stem N; Acorn w/cap N; Acorn w/o cap N; Acorn cap N

PHY 10842

Leaf N; Stem N; Acorn cap Y; Nut N

Quercus cf. *prinoides* Willd. var. *acuminata* (Michx.) Gl.

PHY 10613

Leaf w/petiole N; Small branch Y; Large branch Y

Gramineae

Unknown species

PHY 10788

Leaf Y; Sheath Y; Stem Y; Root Y

Arundinaria gigantea (Walt.) Chapm.

PHY 10618

Leaf Y; Leaf blade Y; Sheath Y; Leaf sheath Y; Stem w/node Y; Stem Y; Small stem Y;
Root Y

PHY 10648

Leaf blade Y; Leaf Sheath Y; Stem Y; Root ?

Digitaria ? *sanguinalis* (L.) Scop.

PHY 10621

Leaf Y; Sheath Y; Stem Y; Inflorescence Y; Root Y

Elymus virginicus L. var. *virginicus* f. *hirsutiglumis* (Scribn.) Fern.

Phy 10833

Leaf Y; Sheath Y; Stem Y; Inflorescence Y; Root Y

? *Muhlenbergia* sp.

PHY 10620

Leaf Y; Stem and sheath Y; Root Y

cf. *Panicum* sp.

PHY 10622

Leaf Y; Sheath Y; Stem Y; Root Y

Panicum dichotomum L.

PHY 10791

Leaf Y; Stem Y; Root Y

PHY 10824

Leaf w/sheath Y; Small leaf + tiny inflorescences Y; Stem Y; Root Y

Panicum lanuginosum Ell. var. *implicatum* (Scribn.) Fern.

PHY 10840

Leaf Y; Stem Y; Inflorescence Y; Root Y

Poa sp.

PHY 10787

Leaf Y; Sheath Y; Stem Y; Root ?

Setaria glauca (L.) Beauv.

PHY 10652

Leaf blade Y; Sheath Y; Stem Y; Inflorescence Y; Root Y

Spenopholis intermedia Rydb.

PHY 10790

Leaf Y; Sheath Y; Stem Y; Inflorescence Y; Root Y

Uniola latifolia Michx.

PHY 10609

Leaf Y; Leaf sheath Y; Stem Y; Inflorescence Y; Root Y

Haloragidaceae

Myriophyllum heterophyllum Michx.

PHY 10835

Leaf N; Stem N; Root N

Hamamelidaceae

Liquidambar styraciflua L.

PHY 10653

Seed Y

Hypericaceae

Hypericum mutilum L. var. *parviflorum* (Willd.) Fern.

PHY 10837

Leaf N; Stem N; Inflorescence N; Root N

Hypericum punctatum L. var. *punctatum*

PHY 10841

Leaf N; Stem Y; Inflorescence Y

Juglandaceae

Carya sp.

PHY 10654

Seed Y

Carya cordiformis (Wang.) K. Koch

PHY 10615

Leaflet Y; Petiole Y; Small branch Y; Large branch Y

Carya ovalis (Wang.) Sarg.

PHY 10650

Leaflet Y; Petiole N; Stem N

Juglans nigra L.

PHY 10607

Leaflet Y; Petiole stem Y; Small branch Y; Large branch Y

Labiatae

Unknown species

PHY 10817

Leaf Y; Stem ?; Root ?

Blephilia hirsuta (Pursh) Benth. var. *hirsuta*

PHY 10596

Leaf N; Stem N; Inflorescence ?; Root ?

Monarda Russeliana Nutt.

PHY 10827

Leaf Y; Stem Y; Inflorescence ?; Root Y

Prunella vulgaris L.

PHY 10816

Leaf Y; Stem ?; Inflorescence ?; Root ?

Pycnanthemum tenuifolium Schrad.

PHY 10830

Leaf Y; Stem Y; Inflorescence ?; Root ?

Lauraceae

Lindera Benzoin (L.) Blume

PHY 10848

Leaf Y; Large stem Y; Small stem Y

Sassafras albidum (Nutt.) Nees var. *albidum*

PHY 10616

One-lobed Leaf w/petiole ?; Two-lobed Leaf w/petiole ?; Three-lobed Leaf w/petiole ?;
Small branch ?; Large branch ?

Sassafras albidum (Nutt.) Nees var. *molle* (Raf.) Fern.

PHY 10634

One-lobed leaf ?; Two-lobed leaf ?; Three-lobed Leaf ?; Small stem ?; Large stem ?;
Root ?

PHY 10772

One-lobed leaf N; Two-lobed leaf N; Three-lobed leaf N; Small branch N;
Large branch N

Leguminosae

Amorpha fruticosa L.

PHY 10625

Leaflet Y; Petiole Y; Stem or small branch Y

Apios americana Medic.

PHY 10821

Leaflet Y; Stem Y; Root ?

cf. *Baptisia* sp.

PHY 10794

Leaflets N; Stem N

Cassia fasciculata Michx. var. *fasciculata* f. *fasciculata*

PHY 10776

Compound leaf ?; Stem ?; Inflorescence ?; Root ?

Cassia fasciculata Michx. var. *robusta* (Pollard) Macbr.

PHY 10597

Compound leaf ?; Stem N; Root N

Cercis canadensis L. var. *canadensis*

PHY 10810

Leaf N; Small branch N; Pod and seed N

Desmodium sp.

PHY 10815

Leaflets Y; Stem Y; Root ?

PHY 10829

Leaf Y; Stem N; Root ?

Desmodium nudiflorum (L.) DC. f. *nudiflorum*

PHY 10633

Leaflet Y; Petiole N; Stem Y; Seed N; Root Y

Phy 10826

Leaf Y; Stem Y; Inflorescence Y

Gleditsia triacanthos L. var. *triacanthos*

PHY 10844

Leaflet Y; Petiole Y; Stem Y; Seed N; Seed pod ?

Lespedeza cuneata (Dumont) G. Don

PHY 10813

Leaf Y; Stem Y; Root ?

Trifolium pratense L.

PHY 10778

Leaflets N; Stem N; Inflorescence N; Root N

Liliaceae

Smilax rotundifolia L.

PHY 10599

Leaf w/petiole N; Stem w/thorn ?; Tendrils ?; Root ?

Magnoliaceae

Liriodendron tulipifera L.

PHY 10812

Leaf N; Stem ?; Inflorescence N

Malvaceae

Hibiscus lasiocarpus Cav.

PHY 10783

Leaf w/petiole N; Stem small N; Medium stem N; Large stem N; Inflorescence N;
Immature inflorescence N; Root N

Melastomataceae

Rhexia virginica L. var. *virginica*

PHY 10775

Leaf ?; Stem ?; Inflorescence ?; Root ?

Menispermaceae

Menispermum canadense L.

PHY 10649

Leaf Y; Petiole N; Stem N

Najadaceae

Potamogeton nodosus Poir.

PHY 10836

Leaf N; Stem ?; Root ?

Nymphaeaceae

Nuphar luteum (L.) Sibth. & Sm.

PHY 10627

Leaf ?; Stem ?; Root ?

PHY 10846

Root ?

Nuphar luteum (L.) Sibth. & Sm. subsp. *macrophyllum* (Small) Beal

PHY 10617

Leaf ?; Stem ?; Inflorescence ? Root (x-section and adventitious) ?

Orchidaceae

Unknown species

PHY 10814

Leaf N; Root N

Oxalidaceae

Oxalis stricta L.

PHY 10624

Leaf w/petiole Y; Stem Y; Inflorescence Y; Root Y

Phytolaccaceae

Phytolacca americana L. Pokeweed

PHY 10784

Large leaf Y; Main Stem N; Inflorescence Stem N; Immature Inflorescence N; Seed ?;
Inflorescence green N; Inflorescence purple N

Pinaceae

Pinus echinata Mill.

PHY 10635

Needle Y; Small branch Y; Large branch Y

PHY 10640

Needle Y; Stem ?; Bark Y; Cone Y

PHY 10643

Needle Y; Branch Y; Bark flakes Y; Open cone ?; Unopened cone ?

Plantaginaceae

Plantago lanceolata L.

PHY 10832

Leaf N; Inflorescence N; Root N

Plantago Rugelii Dcne. var. *Rugelii* Rugel

PHY 10770

Leaf N; Stem N; Seed w/wo stem N; Root N

Platanaceae

Plantanus occidentalis L. var. *occidentalis* f. *occidentalis*

PHY 10610

Leaf w/petiole Y; Small branch Y; Large branch Y; Seed Y; Bract ?

Polygonaceae

Polygonum sp.

PHY 10626

Leaf Y; Stem Y; Root Y

Polygonum hydropiperoides Michx.

PHY 10628

Leaf Y; Stem w/node ?; Stem ?; Inflorescence ?; Root Y

Polygonum virginianum L. var. *virginianum*

PHY 10603

Leaf Y; Stem N; Root (with adventitious roots) ?

Ranunculaceae

Anemone virginianan L.

PHY 10612

Leaflet ?; Stem Y; Root Y

Rosaceae

Agrimonia rostellata Wallr.

PHY 10623

Leaflet Y; Petiole Y; Stem Y; Root Y

Fragaria virginiana Duchesne var. *illinoensis* (Prince) Gray

PHY 10804

Leaf Y; Stem N; Root ?

Rubus sp.

PHY 10595

Leaflet ?; Stem ?; Root ?

Rubiaceae

Cephalanthus occidentalis L. var. *occidentalis* f. *occidentalis*

PHY 10629

Leaf w/petiole N; Small branch N; Large branch Y; Inflorescence Y; Root Y

PHY 10645

Leaf w/petiole N; Stem Y; Large stem Y; Inflorescence N

Rutaceae

Ptelea trifoliata L. var. *trifoliata* f. *trifoliata*

PHY 10808

Leaflet and petiole N; Small branch N; Large branch N; Inflorescence N

Saururaceae

Saururus cernuus L.

PHY 10614

Leaf w/petiole Y; Stem ?; Root Y

Saxifragaceae

Hydrangea arborescens L. var. *Deamii* St. John f. *Deamii*

PHY 10782

Leaf N; Small leaves with inflorescence N; Small stem N; Large stem N; Inflorescence N;
Root N

Penthorum sedoides L.

PHY 10831

Leaf Y; Stem ?; Inflorescence N; Root ?

Scrophulariaceae

Gerardia grandiflora Benth.

PHY 10651

Leaf ?; Small stem N; Large stem ?; Seed N; Seed coat Y; Root Y

Sparganiaceae

Sparganium americanum Nutt.

PHY 10801

Leaf Y; Stem N; Inflorescence N; Root N

Taxodiaceae

Taxodium distichum (L.) Rich. var. *distichum* f. *distichum*

PHY 10592

Compound leaf N; Stem ?

PHY 10593

Bark ?; Wood material ?

PHY 10594

Seed N

Tiliaceae

Tilia americana L.

PHY 10606

Leaf (w/central vein and petiole) Y; Stem Y; Branch Y

Ulmaceae

Ulmus cf. *alata* Michx.

PHY 10792

Leaf Y; Small branch Y; Large branch Y

Umbelliferae

Daucus Carota L. f. *Carota*

PHY 10779

Leaf N; Stem N; Inflorescence N; Root N

Violaceae

Viola sp.

PHY 10809

Leaf N; Stem N; Root N

Viola lanceolata L. var. *lanceolata*

PHY 10785

Leaf N; Stem Y; Root ?

Vitaceae

Parthenocissus quinquefolia (L.) Planch f. *quinquefolia*

PHY 10605

Leaflet ?; Stem Y; Root (at bulb w/main root and adventitious roots) ?

Vitis sp.

PHY 10631

Leaf Y; Petiole Y; Stem N; Tendril N; Large stem N

PHY 10838

Leaf ?; Stem Y; Bark N; Tendril Y; Stem w/o bark N

Vitis vulpina

PHY 10644

Leaf N; Stem Y; Tendril Y

APPENDIX V: POTENTIAL NATIVE FOOD AND BEVERAGE PLANTS OF THE SOUTHEAST MISSOURI OZARKS

The following list of plants is a compilation of the potential native food and beverage plants currently available in the vicinity of the study locality. It includes those plants listed as edible and native by Phillips (1979), King (1976), and Steyermark (1963). Scientific and colloquial names of the plants are listed alphabetically by the scientific name, following the taxonomy of Steyermark (1963). Numbers following abbreviations indicate page references: Phil. = Phillips, J. (1979), King = King, F.B. (1976), and Sty. = Steyermark, J.A. (1963).

Acacia angustissima (Mill.) Ktze. var. *hirta* (Nutt.) Robinson, Prairie acacia, Sty. 869

Acer spp., Sty. 1011, King 251, Phil. 134

Acer Negundo L., Box elder, Sty. 1018, King 251, Phil. 134

Acer nigrum Michx., Black maple, Sty. 1014, King 251, Phil. 134

Acer rubrum L., Red maple, Sty. 1016, King 251, Phil. 134

Acer saccharinum L., Silver maple, Sty. 1018, King 251, Phil. 134

Acer saccharum Marsh, Sugar maple, Sty. 1012, King 251, Phil. 134

Aesculus glabra Willd., Ohio buckeye, Sty. 1019, King 251

Aesculus Pavia L., Red buckeye, Sty. 1022

Agastache nepetoides (L.) Ktze., Yellow giant hyssop, Sty. 1276, King 251

Alisma Plantago-aquatica L., Water plantain, Sty. 58, King 251

Allium canadense L., Wild garlic, Sty. 428, King 251, Phil. 118

Allium cernuum Roth, Nodding wild onion, Sty. 429, Phil. 118

Allium mutabile Michx., Wild onion, Sty. 428, King 251, Phil. 118

Allium stellatum Fraser, Wild onion, Sty. 429, King 251, Phil. 118

Allium tricoccum Ait., Wild leek, Sty. 425, Phil. 118

Amaranthus spp., Sty. 620, King 251

Amaranthus graecizans L., Tumbleweed, Sty. 624, King 251
Amaranthus hybridus L., Green amaranth, Sty. 623, King 251, Phil. 105
Amaranthus retroflexis L., Rough green amaranth, Sty. 623, King 251
Ambrosia trifida L., Horse weed, Sty. 1538, King 251
Amelanchier arborea (Michx. f.) Fern., Shadbush, Sty. 800, King 251, Phil. 20
Amorpha canescens Pursh, Lead plant, Sty. 901, King 251
Amphicarpa bracteata (L.) Fern., Hog peanut, Sty. 953, King 251
Anemonella thalictroides (L.) Spach, Rue anemone, Sty. 704, King 251
Antennaria neglecta Greene, Pussy's toes, Sty. 1532, Phil. 50
Antennaria plantaginifolia (L.) Hook., Pussy's toes, Sty. 1532
Apios americana Medic., Groundnut, Sty. 947, King 251, Phil. 129
Aquilegia canadensis L., Columbine, Sty. 678, King 251
Aralia racemosa L., Spikenard, Sty. 1112, King 251
Aralia spinosa L., Hercules' club, Sty. 1112
Arisaema atrorubens (Ait.) Blume, Jack-in-the-pulpit, Indian turnip, Sty. 383, King 251, Phil. 85
Arisaema Dracontium (L.) Schott, Green dragon, Sty. 384, King 251, Phil. 85
Artemisia caudata Michx. var. *caudata*, Wild wormwood, Sty. 1607
Artemisia ludoviciana Nutt., White sage, Sty. 1610, King 251
Arundinaria gigantea (Walt.) Champm., Cane, Sty. 81
Asarum canadense L., Wild ginger, Sty. 572, King 251, Phil. 114
Asclepias incarnata L., Swamp milkweed, Sty. 1204, King 252, Phil. 138
Asclepias syriaca L. var. *kansana* (Vail) Palmer and Styerm., Common milkweed, Sty. 1206, King 252, Phil. 136
Asclepias tuberosa L., Butterfly weed, Sty. 1203
Asimina triloba (L.) Dunal, Pawpaw, Sty. 671, King 252, Phil. 115
Astragalus caryocarpus Ker, Ground plum, Sty. 908

Astragalus mexicanus A. DC. var. *trichocalyx* (Nutt.) Fern., Ground plum, Sty. 910, King 252

Blephilia ciliata (L.) Britt. var. *ciliata*, Ohio horse mint, Sty. 1294, King 252, Phil. 141

Blephilia hirsuta (Pursh) Benth. var. *hirsuta*, Wood mint, Sty. 1294, Phil. 141

Boehmeria cylindrica (L.) Sw., False nettle, Sty. 568, Phil. 100

Bumelia lanuginosa (Michx.) Pers. var. *albicans* Sarg., Chittim-wood, Sty. 1172, King 252

Callirhoe digitata Nutt. var. *digitata*, Fringed poppy mallow, Sty. 1050, King 252

Campanula americana L., Tall Bellflower, Sty. 1430, King 252

Cardamine bulbosa (Schreb.) BSP., Spring Cress, Sty. 748, King 252, Phil. 17

Cardamine pensylvanica Muhl. var. *pensylvanica*, Bitter-cress, Sty. 750, Phil. 17

Carpinus caroliniana Walt. var. *virginia* (Marsh.) Fern., Blue beech, Sty. 527, King 252

Carya spp., Sty. 512, King 252

Carya aquatica (Michx. f.) Nutt., Water hickory, Sty. 516, King 252

Carya cordiformis (Wang.) K. Koch, Bitternut hickory, Sty. 516, King 252

Carya illinoensis (Wang.) K. Koch, Pecan, Sty. 514, King 252

Carya laciniosa (Michx.) Loud, Big shellbark hickory, Sty. 518, King 252, Phil. 93

Carya ovalis (Wang.) Sarg., False shagbark, Sty. 521, King 252

Carya ovata (Mill.) K. Koch, Shagbark hickory, Sty. 517, King 252, Phil. 93

Carya texana Buckl., Black hickory, Sty. 522, King 252

Carya tomentosa Nutt., Mockernut hickory, Sty. 518, King 252

Castanea dentata (Marsh.) Borkh., Chestnut, Sty. 530

Castanea ozarkensis Ashe., Ozark chinquapin, Sty. 531

Ceanothus americanus L., New Jersey tea, Sty. 1030, King 252, Phil. 34

Ceanothus ovatus Desf., Redroot, Sty. 1030, King 252

Celtis occidentalis L., Hackberry, Sty. 558, King 252, Phil. 97

Cenchrus longispinus (Hack.) Fern., Sandbur, Sty. 240, King 252

Cercis canadensis L. var. *canadensis*, Eastern redbud, Sty. 876, King 252, Phil. 125

Chenopodium album L., Pigweed, Sty. 611, King 252, Phil. 103
Cirsium altissimum (L.) Spreng., Tall thistle, Sty. 1622, King 253
Claytonia virginica L., Spring beauty, Sty. 636, King 253, Phil. 9
Comandra Richardsiana Fern., Bastard toadflax, Sty. 572, King 253
Commelina diffusa Burm., Day-flower, Sty. 398, King 253, Phil. 148
Commelina erecta L., Day-flower, Sty. 400, Phil. 148
Corylus americana Walt., Hazelnut, Sty. 524, King 253
Crataegus mollis (T. and G.) Scheele, Summer haw, Sty. 816, King 253, Phil. 21
Crataegus pruinosa (Wendl.) K. Koch, Frosted haw, Sty. 818, King 253
Crataegus uniflora Muenchh., One flower hawthorne, Sty. 804, King 253
Cryptotaenia canadensis (L.) DC., Honewort, Sty. 1134, King 253, Phil. 36
Cunila origanoides (L.) Britt., Dittany, Sty. 1300, King 253, Phil. 141
Cyperus esculentus L., Yellow nut grass, Sty. 264, King 253
Cyperus ovularis (Michx.) Torr., Hedgehog club rush, Sty. 270
Cyperus refractus Engelm., Umbrella sedge, Sty. 267
Cypripedium Calceolus L., Small yellow lady-slipper, Sty. 470, King 253
Daucus pusillus Michx., Wild carrot, Sty. 1148, King 253
Dentaria laciniata Muhl., Toothwort, Sty. 751, King 253
Dicliptera brachiata (Pursh) Spreng., Sty. 1380, King 253
Diospyros virginiana L., Persimmon, Sty. 1174, King 253, Phil. 39
Echinochloa muricata (Beauv.) Fern. var. *microstachya* Wieg., Barnyard grass, Sty. 236, King 253
Enchinochloa muricata (Beauv.) Fern. var. *occidentalis* Weig., Barnyard grass, Sty. 234, King 253
Eclipta alba (L.) Hassk., Yerba de Tajo, Sty. 1554, King 253
Elymus canadensis L., Canada wild rye, Sty. 130, King 253
Elymus glaucus Buckley, Blue wild rye, Sty. 128, King 254
Epilobium coloratum Biehler, Willow herb, Sty. 1099, King 254

Equisetum hyemale L., Winter scouring rush, Sty. 14
Erigenia bulbosa (Michx.) Nutt., Harbinger of spring, Sty. 1129, King 254
Erythronium albidum Nutt., White dog-tooth violet, Sty. 434, King 254
Erythronium americanum Ker, Adder's tongue, Sty. 433, King 254
Euonymus atropurpureus Jacq., Wahoo, Sty. 1008
Festuca octoflora Walt., Six-weeks fescue, Sty. 89, King 254
Fragaria virginiana Duchesne var. *illinoensis* (Prince) Gray, Wild strawberry, Sty. 824, King 254, Phil. 22
Galium Aparine L., Cleavers, Sty. 1389, King 254, Phil. 44
Galium arkansanum Gray, Bedstraw, Sty. 1392, Phil. 44
Galium boreale L. var. *hyssopifolium* (Hoffm.) DC., Northern bedstraw, Sty. 1392, Phil. 44
Galium circaeazans Michx., Wild licorice, Sty. 1390, Phil. 44
Galium concinnum T. and G., Bedstraw, Sty. 1395, Phil. 44
Galium obtusum Bigel. var. *obtusum*, Bedstraw, Sty. 1394, Phil. 44
Galium pilosum Ait., Hairy bedstraw, Sty. 1390, Phil. 44
Galium tinctorium L. var. *tinctorium*, Bedstraw, Sty. 1394, Phil. 44
Galium triflorum Michx. var. *triflorum*, Sweet scented bedstraw, Sty. 1389, Phil. 44
Galium virgatum Nutt., Bedstraw, Sty. 1389, Phil. 44
Gleditsia triacanthos L., Honey locust, Sty. 873, King 254
Glyceria striata (Lam.) Hitchc., Fowl meadow grass, Sty. 96, King 254
Gymnocladus dioica (L.) K. Koch., Kentucky coffee tree, Sty. 872, King 254, Phil. 31
Hedeoma pulegioides (L.) Pers., Pennyroyal, Sty. 1296, King 254, Phil. 141
Helianthus divaricatus L. var. *divaricatus*, Sunflower, Sty. 1572
Helianthus laetiflorus Pers. var. *rigidus* (Cass.) Fern., Stiff sunflower, Sty. 1570
Helianthus Maximiliana Schrad., Maximilian sunflower, Sty. 1572, King 254, Phil. 76
Helianthus petiolaris Nutt., Prairie sunflower, Sty. 1569, King 254, Phil. 76

Helianthus tuberosus L., Jerusalem artichoke, Sty. 1575, King 254, Phil. 77
Hibiscus lasiocarpus Cav., Rose mallow, Sty. 1053, Phil. 133
Hibiscus militaris Cav., Rose mallow, Sty. 1054, Phil. 133
Humulus Lupulus L., Hops, Sty. 566, King 254
Hydrolea uniflora Raf., Waterleaf, Sty. 1240
Hydrophyllum appendiculatum Michx., Woollen breeches, Sty. 1236, King 254
Hydrophyllum canadense L., Broadleaf waterleaf, Sty. 1234
Hydrophyllum virginianum L. var. *virginianum*, Waterleaf., Sty. 1234, King 255
Ipomoea pandurata (L.) G.F.W. Mey., Wild potato vine, Sty. 1216, King 255
Iva ciliata Willd., Marsh elder, Sty. 1536
Juglans cinerea L., Butternut, Sty. 511, King 255
Juglans nigra L., Walnut, Sty. 510, King 255, Phil. 91
Lactuca canadensis L., Wild lettuce, Sty. 1639
Lactuca floridana (L.) Gaertn., Wild lettuce, Sty. 1642, King 255, Phil. 157
Lactuca Scariola L., Prickly lettuce, Sty. 1638
Laportea candensis (L.) Gaud., Wood nettle, Sty. 568, Phil. 100
Lepidium virginicum L. var. *virginicum*, Pepper grass, Sty. 737, King 255, Phil. 14
Lilium michiganese Farw., Michigan lily, Sty. 432, King 255
Lindera Benzoin (L.) Blume, Spice bush, Sty. 718, King 255, Phil. 64
Lithospermum caroliniense (Walt.) MacM., Puccoon, Sty. 1248
Lithospermum incisum Lehm., Yellow puccoon, Sty. 1247, King 255
Mentha arvensis L., Field mint, Sty. 1304
Monarda fistulosa L., Wild bergamot, Sty. 1291, King 255, Phil. 141
Monarda Russeliana Nutt., Horsemint, Sty. 1290, King 255, Phil. 141
Monotropa uniflora L., Indian pipe, Sty. 1156, King 255
Morus rubra L., Red mulberry, Sty. 562, King 255, Phil. 98

Nasturtium officinale R. Br., Water cress, Sty. 757, Phil. 16
Nelumbo lutea (Willd.) Pers., American lotus, Sty. 668, King 255, Phil. 61
Nuphar luteum (L.) Sibth. and Sm., Yellow pond lily, Sty. 665, King 255
Nymphaea odorata Ait., Fragrant water lily, Sty. 666
Oenothera biennis L., Evening primrose, Sty. 1100, King 255
Onoclea sensibilis L., Sensitive fern, Sty. 27, King 255
Opuntia compressa (Salisb.) Macbr., Prickly pear, Sty. 1084, King 255, Phil. 70
Oryzopsis racemosa (Sm.) Ricker, Black-seeded mountain rice, Sty. 168
Osmorhiza Claytoni (Michx.) Clarke., Sweet cicely, Sty. 1126, King 255
Osmorhiza longistylis (Torr.) DC., Anise root, Sty. 1126, King 255
Osmunda cinnamomea L., Cinnamon fern, Sty. 20, King 256
Oxalis stricta L., Yellow wood sorrel, Sty. 960, Phil. 131
Oxalis violacea L., Violet wood sorrel, Sty. 959, King 256, Phil. 131
Panax quinquefolius L., Ginseng, Sty. 1114, King 256, Phil. 109
Panicum spp., Panic grass, Sty. 206, King 256
Panicum agrostoides Spreng., Panic grass, Sty. 229, King 256
Panicum capillare L., Witch grass, Sty. 226, King 256
Panicum dichotomum L., Panic grass, Sty. 213, King 256
Panicum lanuginosum Ell., Panic grass, Sty. 214, King 256
Panicum oligosanthos Schultes, Small panic grass, Sty. 220, King 256
Panicum virgatum L. var. *virgatum*, Switch grass, Sty. 228, King 256
Parthenocissus quinquefolia (L.) Planch., Virginia creeper, Sty. 1034, King 256
Passiflora incarnata L., Maypops, Sty. 1083, King 256
Pedicularis canadensis L., Wood bentony, Sty. 1366, King 256
Petalostemon candidum (Willd.) Michx., White prairie clover, Sty. 900, King 256
Petalostemon purpureum (Vent.) Rydb., Purple prairie clover, Sty. 900, King 256

Phalaris caroliniana Walt., Canary grass, Sty. 188, King 256
Phaseolus polystachios (L.) BSP. var. *polystachios*, Wild bean, Sty. 948, King 256
Physalis spp., Ground cherry, Sty. 1314, King 256, Phil. 71
Physalis angulata L., Ground cherry, Sty. 1320, King 256
Physalis heterophylla Nees. var. *heterophylla*, Ground cherry, Sty. 1317, King 256
Physalis longifolia Nutt., Ground cherry, Sty. 1318, King 256
Physalis missouriensis Mackenz. and Bush, Ground cherry, Sty. 1321, King 256
Physalis pubescens L., Ground cherry, Sty. 1321, King 256
Physalis virginiana Mill., Ground cherry, Sty. 1317, King 256
Phytolacca americana L., Pokeweed, Sty. 630, King 256, Phil. 6
Pilea pumila (L.) Gray., Clearwood, Sty. 570, King 256, Phil. 100
Plantago cordata Lam., Heartleaf plantain, Sty. 1381, King 256, Phil. 42
Plantago Rugelii Dcne. var. *Rugelii*, Plantain, Sty. 1382, King 256, Phil. 42
Platanus occidentalis L., Sycamore, Sty. 789, King 256
Podophyllum peltatum L., May apple, Sty. 710, King 256, Phil. 12
Polanisia dodecandra (L.) DC. subsp. *dodecandra*, Clammy-weed, Sty. 769, King 256
Polygonatum biflorum (Walt.) Ell. f. *biflorum*, Solomon's seal, Sty. 442, King 257
Polygonatum canaliculatum (Muhl.) Pursh, Solomon's seal, Sty. 442, King 257, Phil. 4
Polygonum aviculare L., Knotweed, Sty. 586, King 257
Polygonum Hydropiper L., Water pepper, Sty. 591, Phil. 5
Polygonum pensylvanicum L., Pinkweed, Sty. 590, King 257
Polygonum scandens L., False buckwheat, Sty. 597
Pontederia cordata L., Pickerel-weed, Sty. 401, King 257
Populus deltoides Marsh., Cottonwood, Sty. 507, King 257
Potamogeton spp., Pondweed, Sty. 50, King 257
Potamogeton amplifolius Tuckerm., Pondweed, Sty. 54, King 257

Potamogeton diversifolius Raf., Pondweed, Sty. 54, King 257

Potamogeton epihydrus Raf. var. *Nuttallii* (C. and S.) Fern., Pondweed, Sty. 54, King 257

Potamogeton foliosus Raf., Pondweed, Sty. 52, King 257

Potamogeton illinoensis Morong., Shining pondweed, Sty. 55, King 257

Potamogeton nodosus Poir., Pondweed, Sty. 55, King 257

Potamogeton pectinatus L., Fennel-leaved pondweed, Sty. 52, King 257

Potamogeton pulcher Tuckerm., Spotted pondweed, Sty. 55, King 257

Potamogeton pusillus L., Pondweed, Sty. 54, King 257

Potentilla simplex Michx., Cinquefoil, Sty. 827, Phil. 67

Prunella vulgaris L., Self-heal, Sty. 1279, King 257, Phil. 151

Prunus spp., Wild plum, Sty. 856, King 257

Prunus americana Marsh., Wild plum, Sty. 860, King 257

Prunus angustifolia Marsh var. *angustifolia*, Chicksaw plum, Sty. 861, King 257

Prunus hortulana Bailey, Wild goose plum, Sty. 860, King 257

Prunus mexicana S. Wats., Big tree plum, Sty. 858, King 257

Prunus Munsoniana Wright and Hedrick, Wild goose plum, Sty. 860, King 257

Prunus serotina Ehrh., Black cherry, Sty. 862, Phil. 67, King 257

Prunus virginiana L., Choke cherry, Sty. 862, King 257

Psoralea esculenta Pursh, Prairie turnip, Sty. 897, King 257

Pteridium aquilinum (L.) Kuhn, Bracken fern, Sty. 21, King 257

Pycnanthemum tenuifolium Schrad., Slender mountain mint, Sty. 1299, Phil. 141

Pyrus coronaria L., Wild crab, Sty. 799

Pyrus ioensis (Wood) Bailey, Wild crab, Sty. 799, King 257, Phil. 121

Quercus spp., Oak, Sty. 532, King 257, Phil. 95

Quercus alba L., White oak, Sty. 535, King 257

Quercus bicolor Willd., Swamp white oak, Sty. 538, King 257

Quercus coccinea Muenchh., Scarlet oak, Sty. 547, King 257
Quercus falcata Michx., Spanish oak, Sty. 544, King 257
Quercus imbricaria Michx., Shingle oak, Sty. 540, King 257
Quercus lyrata Walt., Overcup oak, Sty. 536, King 257
Quercus macrocarpa Michx., Bur oak, Sty. 537, King 257
Quercus marilandica Muenchh., Black Jack oak, Sty. 543, King 257
Quercus Michauxii Nutt., Basket oak, Sty. 538, King 257
Quercus nigra L. var. *nigra* f. *nigra*, Water oak, Sty. 543, King 257
Quercus Nuttallii Palmer, Nuttall's oak, Sty. 551, King 257
Quercus palustris Muenchh., Pin oak, Sty. 550, King 257
Quercus Phellos L. f. *Phellos*, Willow oak, Sty. 542, King 257
Quercus prinoides Willd., Chestnut oak, Sty. 539, King 257
Quercus rubra L., Red oak, Sty. 550, King 257
Quercus Shumardii Buckl., Shumard oak, Sty. 548, King 257
Quercus stellata Wang., Post oak, Sty. 535, King 257
Quercus velutina Lam., Black oak, Sty. 544, King 257
Rhus aromatica Ait., Fragrant sumac, Sty. 1002, King 257, Phil. 68
Rhus copallina L. var. *latifolia* Engler, Dwarf sumac, Sty. 1000, King 257, Phil. 68
Rhus glabra L. var. *glabra*, Smooth sumac, Sty. 1000, King 257, Phil. 68
Ribes Cynosbati L. var. *Cynosbati* f. *Cynosbati*, Prickly gooseberry, Sty. 785, King 257
Ribes missouriense Nutt., Missouri gooseberry, Sty. 785, King 257, Phil. 18
Ribes odoratum Wendland. f., Golden currant, Sty. 785, King 257
Robinia Pseudo-Acacia L., Black locust, Sty. 906, King 257, Phil. 33
Rosa carolina L., Pasture rose, Sty. 852, Phil. 123
Rosa palustris Marsh., Swamp rose, Sty. 852, Phil. 123
Rosa setigera Michx., Prairie rose, Sty. 849, Phil. 123

Rubus spp., Blackberry, Dewberry, Sty. 834, King 258
Rubus allegheniensis Porter, High-bush blackberry, Sty. 840, King 258
Rubus argutus Link, High-bush blackberry, Sty. 841, King 258
Rubus Enslenii Tratt., Dewberry, Sty. 838, King 258
Rubus flagellaris Willd., Dewberry, Sty. 837, King 258, Phil. 28
Rubus invisus (Bailey) Britt., Dewberry, Sty. 838, King 258
Rubus laciniatus Willd., Cut-leaved blackberry, Sty. 836, King 258
Rubus mollior Bailey, High-bush blackberry, Sty. 841, King 258
Rubus occidentalis L. f. *occidentalis*, Black raspberry, Sty. 835, King 258, Phil. 23
Rubus orarius Blanchard, High-bush blackberry, Sty. 841, King 258
Rubus ostryifolius Rybd., High-bush blackberry, Sty. 842, King 258
Rubus pensilvanicus Poir., High-bush blackberry, Sty. 841, King 258, Phil. 25
Rubus trivialis Michx., Southern dewberry, Sty. 837, King 258
Rudbeckia laciniata L. var. *laciniata*, Wild goldenglow, Sty. 1557, King 258
Rumex altissimus Wood, Pale dock, Sty. 578, King 258, Phil. 101
Rumex verticillatus L., Swamp dock, Sty. 578, King 258, Phil. 101
Sagittaria spp., Arrowhead, Sty. 62, Phil. 1
Sagittaria Engelmanniana subsp. *brevirostra* (Mack. and Bush) Bogin, Arrowhead, Sty. 64, Phil. 1
Sagittaria graminea Michx., Arrowhead, Sty. 63, Phil. 1
Sagittaria latifolia Willd., Duck potato, Sty. 64, King 258, Phil. 1
Sagittaria montevidensis Cham. and Schl., subsp. *calycina* (Engelm.) Bogin, Arrowhead, Sty. 62, Phil. 1
Sagittaria rigida Pursh, Stiff arrowhead, Sty. 63, Phil. 1
Salix spp., Willow, Sty. 489, Phil. 90
Salix amygdaloides Anders., Peach-leaved willow, Sty. 494, Phil. 90
Salix caroliniana Michx., Ward's willow, Sty. 494, Phil. 90

Salix eriocephala Michx., Willow, Sty. 498, Phil. 90
Salix humilis Marsh., Prairie willow, Sty. 500, Phil. 90
Salix interior Rowlee, Sandbar willow, Sty. 497, Phil. 90
Salix nigra Marsh., Black willow, Sty. 494, Phil. 90
Salix rigida Muhl., Willow, Sty. 497, Phil. 90
Salix sericea Marsh., Silky willow, Sty. 501, Phil. 90
Sambucus canadensis L., Common elderberry, Sty. 1418, King 258, Phil. 46
Sassafras albidum (Nutt.) Nees, Sassafras, Sty. 717, King 258, Phil. 62
Satureja arkansana (Nutt.) Briq., Calamint, Sty. 1297, King 258
Scirpus acutus Muhl., Great bullrush, Sty. 292
Scirpus validus Vahl var. *creber* Fern., Great bullrush, Sty. 292, King 258
Setaria geniculata (Lam.) Beauv., Prairie foxtail, Sty. 237, King 258
Silphium laciniatum L. var. *laciniatum*, Compass plant, Sty. 1550, King 258
Sium suave Walt. f. *suave*, Water parsnip, Sty. 1136, King 258
Smilacina racemosa (L.) Desf., False Solomom's seal, Sty. 440, King 258, Phil. 3
Smilacina stellata (L.) Desf. var. *stellata*, Starry false Solomom's seal, Sty. 440
Smilax spp., Catbrier, Sty. 448, Phil. 89
Smilax Bona-nox L. var. *Bona-nox*, Catbrier, Sty. 452, King 258, Phil. 89
Smilax ecirrhata (Engelm.) Wats., Carrion-flower, Sty. 451, Phil. 89
Smilax glauca Walt., Greenbrier, Sty. 451, Phil. 89
Smilax herbacea L., Carrion-flower, Sty. 451, King 258, Phil. 89
Smilax pulverulenta Michx., Carrion-flower, Sty. 450, Phil. 89
Smilax rotundifolia L., Greenbrier, Sty. 454, Phil. 89
Smilax tamnoides L. var. *hispida* (Muhl.) Fern., Bristly greenbrier, Sty. 452, King 258, Phil. 87
Solanum americanum Mill., Black nightshade, Sty. 1312, King 258
Solidago missouriensis Nutt., Goldenrod, Sty. 1488, King 259

Solidago odora Ait., Sweet goldenrod, Sty. 1494
Sparganium americanum Nutt., Bur-reed, Sty. 49, King 259
Spiranthes gracilis (Bigel.) Beck, Slender ladies' tresses, Sty. 480, King 259
Spiranthes vernalis Engelm. and Gray, Ladies' tresses, Sty. 480, King 259
Sporobolus spp., Dropseed, Sty. 161, King 259
Sporobolus clandestinus (Biehler) Hitchc., Dropseed, Sty. 165, King 259
Sporobolus cryptandrus (Torr.) A. Gray var. *cryptandrus*, Sand dropseed, Sty. 166, King 259
Sporobolus heterolepis Gray, Prairie dropseed, Sty. 165, King 259
Sporobolus neglectus Nash, Poverty grass, Sty. 162, King 259
Sporobolus vaginiflorus (Torr.) Wood, Poverty grass, Sty. 162, King 259
Staphylea trifolia L., American bladder-nut, Sty. 1010, King 259
Strophostyles helvola (L.) Ell., Wild bean, Sty. 951, Phil. 130
Symphoricarpos orbiculatus Moench f. *orbiculatus*, Coral berry, Sty. 1410, Phil. 110
Tilia americana L., Basswood, Linden, Sty. 1042, King 259, Phil. 35
Tilia heterophylla Vent., White basswood, Sty. 1044
Tradescantia spp., Spiderwort, Sty. 392, King 259
Tradescantia longipes Anderson and Woodson, Wild crocus, Sty. 396, King 259
Tradescantia ohioensis Raf., Spiderwort, Sty. 396, King 259, Phil. 147
Tradescantia subaspera Ker. var. *subaspera*, Spiderwort, Sty. 392, King 259
Tradescantia virginiana var. *virginiana*, Spiderwort, Sty. 394, King 259, Phil. 147
Trillium sessile L., Wake robin, Sty. 444, King 259, Phil. 113
Triosteum perfoliatum L., Common horse gentian, Sty. 1410, King 259, Phil. 117
Typha latifolia L., Common cat-tail, Sty. 46, King 259, Phil. 81
Ulmus rubra Muhl., Slippery elm, Sty. 555, King 259
Uvularia grandiflora Sm., Bellwort, Sty. 424, King 259, Phil. 55
Vaccinium stamineum L., Deerberry, Sty. 1162, King 259

Vaccinium vacillans Torr., Lowbush blueberry, Sty. 1163, King 259, Phil. 38
Valerianella radiata (L.) Dufr., Corn salad, Sty. 1420, King 259, Phil. 49
Verbena hastata L., Blue vervain, Sty. 1258, King 260
Veronica comosa Richter, Water speedwell, Sty. 1354, King 260
Viburnum prunifolium L., Black haw, Sty. 1414, King 260, Phil. 45
Viburnum rufidulum Raf., Southern black haw, Sty. 1415, King 260
Viola spp., Violet, Sty. 1069, Phil. 149
Viola cucullata Ait. var. *cucullata* Marsh, Blue violet, Sty. 1071, Phil. 149
Viola lanceolata L. var. *lanceolata*, Lance-leaved violet, Sty. 1078, Phil. 149
Viola missouriensis Greene, Missouri violet, Sty. 1074, Phil. 149
Viola papilionacea Pursh f. *papilionacea*, Common violet, Sty. 1072, Phil. 149
Viola pedata L., Pansy violet, Sty. 1071, Phil. 149
Viola pensylvanica Michx., Smooth yellow violet, Sty. 1080, Phil. 149
Viola sagittata Ait., Arrow-leaved violet, Sty. 1076, Phil. 149
Viola sororia Willd., Woolly blue violet, Sty. 1074, Phil. 149
Viola striata Ait., Pale violet, Sty. 1080, Phil. 149
Viola triloba Schwein., Three-lobed violet, Sty. 1076, Phil. 149
Viola viarum Pollard, Plains violet, Sty. 1077, Phil. 149
Vitis spp., Grape, Sty. 1035, King 260, Phil. 107
Vitis aestivalis Michx., Summer grape, Sty. 1036, King 260, Phil. 107
Vitis cinerea Engelm., Grayback grape, Sty. 1037, King 260, Phil. 107
Vitis palmata Vahl, Red grape, Sty. 1038, King 260, Phil. 107
Vitis rupestris Scheele, Sand grape, Sty. 1041, King 260, Phil. 107
Vitis vulpina L., Winter grape, Sty. 1040, King 260, Phil. 107
Yucca glauca Nutt. var. *glauca*, Soapweed, Sty. 439
Yucca Smalliana Fern., Spanish bayonet, Sty. 439
Zizania aquatica L. var. *interior* Fasset, Water rice, Indian rice, wild rice, Sty. 193
Zizaniopsis miliacea (Michx.) Döll and Aschers., Water millet, Sty. 193